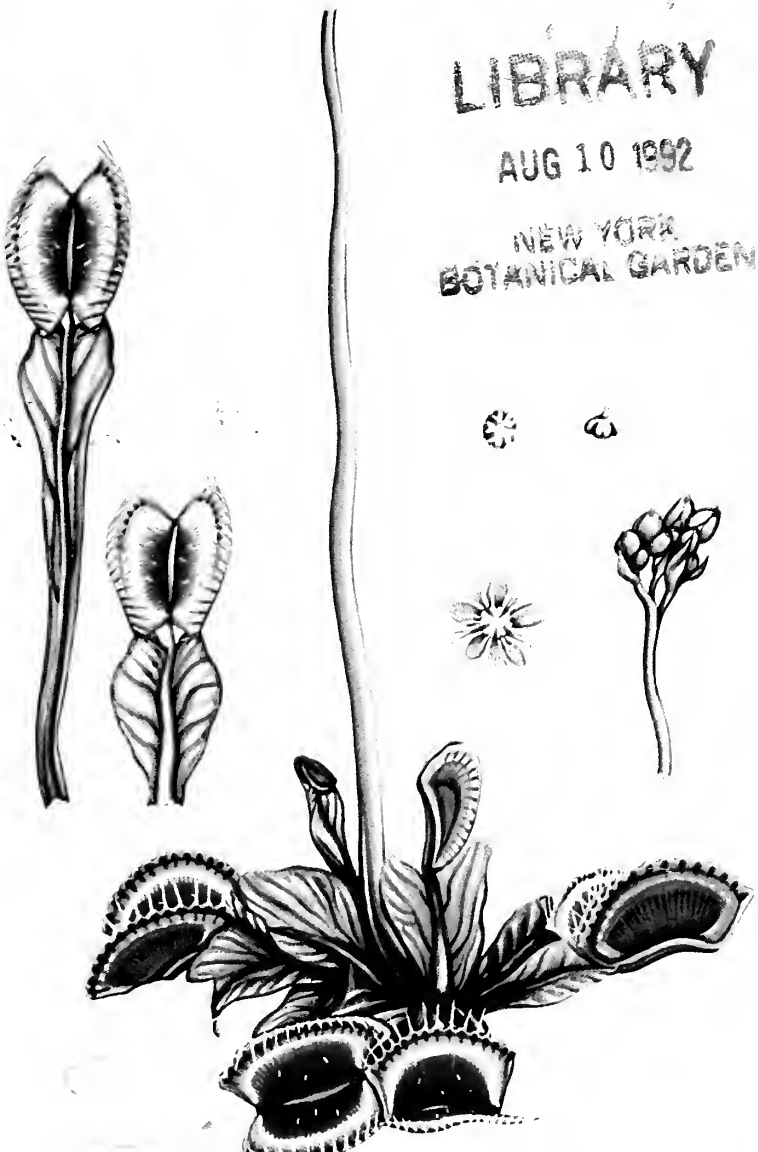
CARNIVOROUS PLANT NEWSLETTER

VOLUME 21, NUMBERS 1 & 2 MARCH & JUNE 1992



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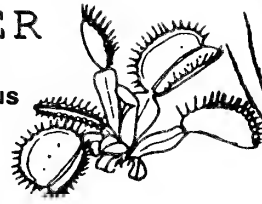
Official Journal of the International Carnivorous Plant Society



Volume 21, Numbers 1 & 2
March & June 1992

Front cover: *Dionaea muscipula* painting (20 x 30) by George Gercama. Please see News & Views page 5.

Rear cover: *Dionaea muscipula* trap with cricket. Photo by and copyrighted by Jerome Wexler. See related article beginning on page 14.

The co-editors of CPN would like everyone to pay particular attention to the following policies regarding your dues to the ICPS.

All Correspondence regarding dues, address changes and missing issues should be sent to ICPS c/o Fullerton Arboretum, CSUF, Fullerton, CA 92634. DO NOT SEND TO THE CO-EDITORS. Checks for subscription and reprints should be made payable to ICPS.

All material for publication, comments and general correspondence about your plants, field trips or special noteworthy events relating to CP should be directed to one of the co-editors. We are interested in all news related to carnivorous plants and rely on the membership to supply us with this information so that we can share it with others.

Views expressed in this publication are those of the authors, not necessarily the editorial staff.

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Editor's Corner

Welcome to another year of CPN. We are presenting another double issue to try and catch up. Thanks for your understanding and support. Please notice that we are again printing the Seed Bank list. Remember that inventory changes may take place from the time I (Leo) get the list and the time that CPN reaches you. Contact Gordon Snelling before you order or list substitutes. Due to time constraints, back orders should not be requested. Be sure to enclose a SASE or 2 IRC's

A note about housekeeping. Articles longer than one CPN page should be on disc if possible. Please send all manuscripts and discs* to Joe Mazrimas. (Address is on inside front cover.) He is the primary editor responsible for sending the article to the appropriate editor for reading and corrections. After this process is complete, it is sent to the Managing Editor for final corrections and preparation for the desktop editor. Pagemaker by Aldus is used for the final formatting of CPN.

**preferred format is Macintosh using Microsoft Word. PC-DOS is acceptable, but we must convert it to Macintosh. Please use 3.5 inch discs. Always enclose a hard copy printed in at least 10 point type or larger. Laser print is preferred if the article must be scanned. Use a sans serif font for less scan error. If you have a dot matrix or manual typewriter, please use a new ribbon.*

Special Announcement

An informal gathering of CP hobbyists will be held at the home of Carl and Sherry Taylor in Lakeport (a small community on Oneida Lake just east of Syracuse, New York) during the weekend of August 7-9, 1992. Carl, Sherry, and Chris Belanger have several events planned, including:

- SLIDE SHOWS BY CHRISTOPH BELANGER and SCOTT BENNETT
- ROUND-TABLE DISCUSSIONS ON PROPAGATION and CHOOSING PROPER SOIL MIXES
- PLANT AND SEED EXCHANGE BY ATTENDEES
- SWIMMING IN ONEIDA LAKE
- BACKYARD BARBEQUES AND BUFFETS
- PLANNING MEETING FOR 1993 CONVENTION

As many of you know, Bill Scholl, Jim Bokowski and several others travelled to Venezuela in February of this year. when they visited Mt. Ilu. Some of the plants they found will be on hand for viewing.

Carl, Cherry, and Chris hope you will also join in a planning meeting to discuss a full-scale convention in August of 1993 at a suitable hotel in the Syracuse area.

Since this gathering is informal, you're welcome to pitch your tent in the backyard and pretend you're collecting in the wilds of Australia (sorry, no poisonous snakes or crocodiles, but mosquitoes will be provided), or there are inexpensive motels within driving distance. Meals will be home-cooked barbeques and buffets, with everyone sharing the preparation and cost.

If you or someone you know would like to attend either the informal gathering or the August, 1993 Convention, please let us know as soon as possible so we can finalize plans. If you write, please include a large, self-addressed, stamped envelope for reply. Contact: CHRISTOPH A. BELANGER (988 Brightwood Drive; Toms River, New Jersey 08753. Phone 908/240-5311) or CARL & SHERRY TAYLOR (2651 Larkin Avenue; Lakeport-Canastota, New York 13032. Phone 315/633-2359).

News & Views

Freddy de Coninck (President of the CPS Drosera; Scientific collaborator; Laboratory of Morphology, Systematics & Ecology of Plants; University of Gent; Ledeganckstraat 35; B-9000; Gent, Belgium) writes:

This year July 9th to 18th, 1991, our CP society "Drosera" and the Botanical Garden of the University of Gent have organized for the first time an International CP Exhibition.

We have received the overwhelming number of 11,300 visitors. All genera of CP were on display, including the rarer ones, such as *Heliamphora* sp., *Brocchinia reducta, Genlisea, Catopsis berteriona, Aldrovanda,* and some Tepui Utrics and *Drosera* species.

Plants were received on loan from several institutions, namely the Botanical Garden of the Universiteit Gent (Belgium), the Botanical Garden of the Vrije Universiteit Amsterdam (Netherlands), the Botanical Garden of the Universität Bonn (Germany), and the Botanical Garden of the Universität Essen (Germany). More contributions were received from many members of our own society, from Y. A. Utz (Switzerland), and from several members of the Dutch society, Carnivora.

The international jury was composed of committee members of each European CP society: Dudley & Margaret Watts (CPS, England), A. Vogel & H. Luhrs (Carnivora, Netherlands), Holger & Anja Herrern (Das Taublatt, Germany), G. Lecointe (Dionée, France), J. Hulab-Novak (CPS, Czechoslovakia), J. De Witte (Drosera, Belgium), Y. A. Utz (Switzerland).

President of the jury was Dr. J.D. Degreef (Liège, Belgium), well known from his many scientific contributions both privately edited and in several CP journals.

This exhibition was unique in several respects, a.o. it was the first time that all European CP Societies were united, and members of these could meet each other and exchange ideas: a fruitful experience!

Ludek Frkal of the Czechoslovak Carnivorous Plant Society (Kuty 1942; 760-01 Zlin; Czechoslovakia) writes:

Founded in January, 1990, the Czechoslovak Carnivorous Plant Society (CCPS) is the organization associating those Czech and Slovak CP hobbyists who have decided to cooperate more closely. The activity of the CCPS comprise the amateur growing, propagating CPs and introducing new and rare species among Czechoslovak CP hobbyists. In collections of 17 CCPS members you can find maybe everything what is just to disposal on the CP market. Of course not everybody has all species: for example there are specialists in pygmy Droseras, tuberous Droseras, Utricularias, etc. The cooperation within the society brings, among others, much more possibilities to buy or exchange something new from abroad. And a species obtained by one member of the Society becomes in one or two seasons attainable to all others. But this is only one side of our activity. The others are publishing materials on CP growing (4 brochures so far), running the seed bank, organizing public exhibitions and meetings, trips into CP localities involving their documentation and restoration, etc. The Society in cooperation with the Masaryk University in Brno would like to start the tradition of annual spring CP sessions in the Brno Botanical Garden with attendance of CP growers from both Czechoslovakia and abroad. This year's session will be held on 29th - 31st May. As for our international activities, last May four members of the CCPS took part in the meeting of the European CP hobbyists in Gent, Belgium. Now the CCPS becomes a member of the ICPS. (Welcome to the ICPS-ed.)

George Gercama (246 Trent Avenue; Winnipeg; Manitoba; CANADA R2K 1E5. Tel.; [h] 204/663-5109; [w] 204/943-1681) writes:

I'm an artist who has been experimenting with botanical illustration for about a year now. My interest in botanical art grew out of a long-standing fascination with succulent flora. While continually searching for new subjects, I became interested in CP, and have experimented a bit with cultivating them from seeds. I have a number of mature *Dionaea muscipula* plants and *Drosera capensis*. The *Dionaea* have made wonderful subjects for a small series of paintings that I completed this spring (1991-ed.). Please see rear cover for painting of *Dionaea*.

Michael Herrera (13679 SW 62nd Street #103; Miami FL 33183; USA) writes:

I am 13 years old and just became a member. I am a good friend of Mr. Clyde Bramblett. I buy all my CP from him. I would really like to thank him for giving me some CP, and especially for taking me out to the Everglades to show me some *Pinguicula pumila* growing wild.

He also wrote that *S. minor* was seen by a friend of Clyde's growing at the northern point of Lake Okeechobee. Also, Michael plans to visit other areas in Florida (Deland and Callahan area) to see CP.

ICPS Seed Bank

(inventory as of 15 May 1992)*

Byblis liniflora

Drosera aliciae, D. binata "Coromandel penn" (6), *D. bin.* "hauki plain" New Zealand, *D. bin.* "Northland, New Zealand", *D. burkeana, D. burmannii, D. capensis* "alba" (13), *D. cap.* (5), *D. cap.* NL (10), *D. cap.* "red leaf", *D. capillaris* "Brasiliensis", *D. capil.* pink (2), *D. capil.* NC (1), *D. collinsae* (2), *D. communis*, D. dielsiana (2), *D. filiformis, D. glanduligera* (5), *D. intermedia, D. int.* "tropical", *D. indica, D. indica* "Qld" (10), *D. linearis* "Bruce Co., Ontario", *D. montana* var. *montana* & *tomentosa, D. rotundifolia, D. rot.* "southern BC", *D. rot.* "New Jersey", *D. spatulata* (13), *D. spath.* "hauraki plain" (7), *D. spath.* "kansai" (2), *D. spath.* "Hong Kong" (5), *D. spath.* "pink flower", *D. villosa* "narrow leaf" Camino do mar, Brasil; Pygmy *Drosera*—> *D. occidentalis occidentalis* (1), *D. pulchella* (2), *D. pygmae* (10); Tuberous *Drosera* —> *D. auriculata* (3), *D. auriculata* "New Zealand", *D. auric.* Melbourne, *D. peltata* "New Zealand".

Pinguicula vulgaris

Utricularia delicatula Waikato NZ, *U. novae-zealandeae* Hauraki plain, *U. longifolia* (8), *U. caerulea, U. hispida* (13), *U. tricolor* (13), *U. livida* (2)

Nepenthes khasiana, N. mirabilis, N. rafflesiana, N. ventricosa, N. ventricosa X (*inermis* X *bongso*)

Sarracenia alata, S. flava, S. flava Eastern Virginia, *S. leucophylla* (almost white), *S. leucophylla, S. purpurea* ssp. ?, *S. purp. purp.* (5), *S. purp.* "BC", *S. purp. purp.* "Quebec", *S. purp. purp.* "Mich" (10), *S. purpurea* X *flava* X self (2), *S.* [*leuco* X (*rubra* X *leuco*)] X *purp.* X *flava*.

**for reference purposes only.* Please send SASE or two IRC's to Gordon Snelling (ICPS Seed Bank; 300 West Carter Drive; Glendora CA 91742; USA) for most recent list. Price is US$1.00/packet. Cash, check, postal money orders, giro/mandat postal, etc.

Seed Bank Donors

August, 1990-February, 1992

Name followed by total number of packets donated during the above time period. Many thanks to those who have supported the Seed Bank. Keep those seeds coming-it is one of the best ways to share CP.

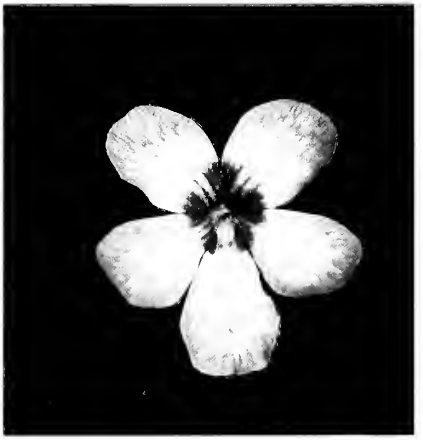
Irena Bukowski 38, Davis Creek Nursery 60, David Crump 20, Cliff Dodd 15, Patrick Dwyer 104, James Fife 83, William Fritsch 15, Robert Gibson 55, Joey Gregore 40, Tom Johnson 129, Tom Kahl 70, Paul Kane 40, Steve Kapa 40, Randy Lamb 180, Rob Maharajh 169, Phill Mann 40, Ric Maulder 315, Bill McLaughlin 110, Joe Mazrimas 89, Barry Meyers-Rice 80, Jim Powell 43, **Fernando Rivadavia 404 top doner**, Gordon Snelling 42, Ivan Snyder 56, R. Schlosser 5, Miloslav Studnika 85, Stan Vangel 17, Charles Webb 20, Bill Webber 14, & Carl Wong 40 for a total of **2418** packets! Inter-Society exchange with the Victorian CPS 40 packets for a grand total of 2458 packets.

Pinguicula "Gina"- a new decorative CP

By Miloslav Studnicka
Botanical Gardens, 460 01 Liberec
Czechoslovakia

The novelty is a hybrid of the species *P. agnata* (male) and *P. zecheri* (female), from different subgenera Isoloba and Pinguicula. The winter rosette is similiar to the one of P. *ag*nata. The summer rosette, consisting of ±13 leaves 8 by 6 cm, measures about 15 cm across. These leaves also resemble *P. ag*nata, but they are a little rolled up on the margins.

Flowers are produced by the winter as well as by the summer rosette. The corolla consists of two lips (bilabiate) and it measures 38 by 33 mm. The corolla tube is 10 mm long. The greenish-yellow spur is 17 mm long. These both are glandular haired and they contain an angle 140°. There are three differently coloured zones from the margins to the centre of the corolla, as seen in the photo. The mouth of the corolla tube is marked by a yellow band. The stigma violet in the center and white in the margins.



Close-up of flower of *Pinguicula X "Gina"* Photo by author.

The hybrid cannot produce any seeds. It is propagated by means of culture "in vitro" in the Botanic Gardens in Liberec (Czechoslovakia) .

The described cultivated variety is dedicated to our colleague Gina N. (Mrs.), who has cooperated in our conservation program of the critically endangered species *Pinguicula bohemica.*

Utricularia nephrophylla

By Christoph A. Belanger
988 Brightwood Dr.
Toms River, N.J. 08753

Utricularia nephrophylla has been the subject of many discussions and the cause of much confusion in my neck of the woods. In the last three years, many plants have surfaces as "new introductions" when in reality they are just the true *U. nephrophylla.* I will try to clear up some of this fruit salad of new non-specific names, and to clear up some of the old misidentifications and to describe this wonderful little epiphytic species that should be in the collection of every Utricularia grower.

This whole mess started for me a few years back when I received my first *Utricularia: U. nephrophylla*. It was a very pretty plant and rather large, much like the one pictured in Adrian Slack's book *Insectivorous Plants and How to Grow Them* on page 126. Unfortunately, this plant is not *U. nephrophylla* as it turns out a few years later, but a smaller variation of *U. reniformis*, according to Peter Taylor's monograph.

About two years after I received my first *Utricularia*, I received my first unidentified species. It was sent to me as *U. sp. 'Roraimae'* and it is known still today under that name. From what I could find out from other growers, German growers "discovered" it in Venezuela, about four years ago. This plant however, that has been known to so many people as *U. sp. 'Roraimae'* is, according to Peter Taylor's monograph, the true *U. nephrophylla.*

About a year later, I received another plant from Brazil, this one identified as *U. nephrophylla*. Since I doubted the true identity of this plant, and Slack's Book seemed to have the correct identifications, and the plant I received did not at all look similar to the picture in this book, I proceeded to give it a non-specific temporary name: *U. sp. 'Rio de Janeiro'*, another name to add to the fruit salad of names. This plant turned out to have the same flowers as *U. sp. 'Roraimae'* except for a small variation in the shape of the corolla. Even so, I thought that the similarities in the flower were close enough to say that the plants were of the same species. There are still heated discussions since the shapes of the leaves were much different: *U. sp. 'Roraimae'* having leaves up to 10 mm wide and *U. sp. 'Rio de Janeiro'* having leaves not even 3 mm wide, the length of the petioles being about equal.

Then, finally, the work of works was published, *The Genus Utricularia*, by Peter Taylor, and correct identifications could be made. *Utricularia sp. 'Roraimae'* and *U. sp. 'Rio de Janeiro'* are both correctly identified as *U. nephrophylla*. The former, as the name implies, from Mt. Roraima, Venezuela, and the later from Rio de Janairo, Brazil.

Now, what does this plant look like? Well it is very pretty, but that does not do too much for its description. The leaves are more or less numerous, through more numerous and more delicate in the form from Rio de Janairo. In the form from Mt. Roraima they are rather kidney shaped with a width of up to 10 mm wide as observed in my own plants. The petiole can be various lengths all depending on how deep the stolons are. In the form from Rio de Janeiro the leaves are almost minute, barely reaching 4 mm in width, and are located on a rather long petiole in comparison to the leaf, about 5 mm long, and the stolons are usually located directly under the surface of the growing medium.

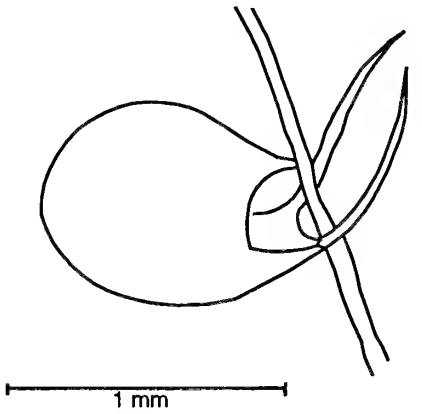
The flowers, even thought they are not exactly large, are very pretty. They are located on an inflorescence which is about 15 to 20 cm long, usually solitary in the form from Mr. Roraima, and up to two in the form from Rio de Janeiro. The flowers in both of these forms are white with two yellow crests situated at the base of the lower lip. The upper lip is ovate, and in my plants, is arched over crests of the lower lip and is about

7 mm long and 5 mm wide. The lower lip in my plants is about 13 m long and about 12 mm wide. The lower lip has two distinct lobes with the spur, which is about 8 mm long, fitting between the two lobes. Taylor mentions a third lobe that may be present directly between the two lateral lobes which is very small, though it is not present at all times and I have not seen it in my own plants.

U. nephrophylla is a very easy plant to grow. It can easily be grown and flowered in a two inch pot since it is not at all a larger grower. I have used several different mediums with equal success, there being mixtures between peat, sand and dried *Sphagnum*, and just pure live *Sphagnum* (for the larger Roraima form). I have grown my plants in 10 gallon tanks with other plants such as *Heliamphora*, and other *Utricularia*, which require warmer temperatures. If the plants are left alone they will reward your patience with a beautiful flower in a short time. After the flower has died, the plant should be divided and it will continue to grow vigorously.

One thing must be clarified, though. As the monograph is read one will notice a discrepancy between the distribution of *U. nephrophylla* in Taylor's monograph and this article. Taylor states that the plant occurs mainly in Brazil, and does not even mention Mt. Roraima or even Venezuela. I am assuming, since it was known as "sp. Roraima", that it was collected there, however, I am not at all sure of this.

Even so, this does not change the fact that I still think it is a very pretty plant and that every serious collector of carnivorous plants should have an example of *U. nephrophylla.*



Utricularia nephrohylla trap

Leaf of *U. nephrophylla* of the Roraima form

Leaf of *U. nephrophylla* of the Rio de Janeiro form

Drawings by
Christoph A. Belanger
April 12, 1991

Focusing on *U. calycifida*—a Variable Species

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Since Peter Taylor's *Utricularia* monograph was published in 1989, learning about the plants in our collections has become much easier. But, since the genus is large and globally widespread, the monograph is technical and expensive and not likely to be in every CPer's library. The result is many CP enthusiasts don't have a way to get to the valuable information in Taylor's work. This is why I'm writing the Focusing on *Utricularia* articles—to present parts of Taylor's work to CPer's. Each installment of this series will be about a different species, a species commonly in cultivation but not illustrated in CPN or other easily accessible sources. I'll try to include enough of a description of each species so you can verify the identification of your plant if you think you're growing it. Space allowing, I'll include a photograph or two. I already have the next few articles planned, but if I hear of much interest in a particular species I can put it in the queue. With 214 species of *Utricularia* described, identifying a plant can be hard. Often a very good (or the best) way may involve microscopic details—the precise nature of the bladders, seed coats and such. I'll avoid these details and spend most of my time on the macroscopic features of the plants—leaves, flowers, or features that can be seen with a magnifying glass. If you grow the plant being discussed, aggressively compare my description to your specimen. Since the time the ancestor of your plant was originally field collected, its descendents may have passed through the hands of many growers, and could have become mislabelled on the journey. Treat the name tags in your pots with a healthy skepticism. It is very helpful if you know the country from which your plant (or its ancestor) was collected. If you have this information for your plants, record it.

Species of *Utricularia* can be variable. Not only can one clone be different from another, but the same plant can produce wildly different leaves and flowers under differing conditions. As an example, consider *U. subulata*—a weed in my CP collection. Sometimes the flowers on my plants are cleistogamous—forever pale white, small and budlike—and are produced on short erect inflorescences. Meanwhile, a *subulata* pot next to it may be producing many brilliant orange-yellow flowers several millimeters long on lanky inflorescences that eventually topple onto other pots. Other pots have plants producing inflorescences of intermediate form. In fact, during the year a plant may change among these forms. Clearly it would be wrong to assign different species or subspecies names to these different growth habits (although this has been mistakenly done in the past, e.g. '*U. cleistogama*'). Remember some species are prone to variation, and just because your plant is producing small flowers instead of large, or lavender flowers instead of lilac, does not mean you have a different species. As I said before, the characteristics that best distinguish one species from another may not be the flowers alone.

The tropical *U. calycifida* is a good example of variability within a species. Before I talk about its variable features, let's look at what most of the forms have in common. The above ground part of the olive green leaves can be large—easily 5 cm or longer, are roughly oval shaped, and are semi-erect or lie on the ground. They are either scattered helter-skelter on the soil surface or arranged in a rosette. The largest are fleshy, and are veined in a complex pattern (Figure 1). While a leaf is growing its veins may be purplish, but this can fade as it matures. The bladders of the plant are small (about 2 mm maximum), and the plant does not produce tubers.

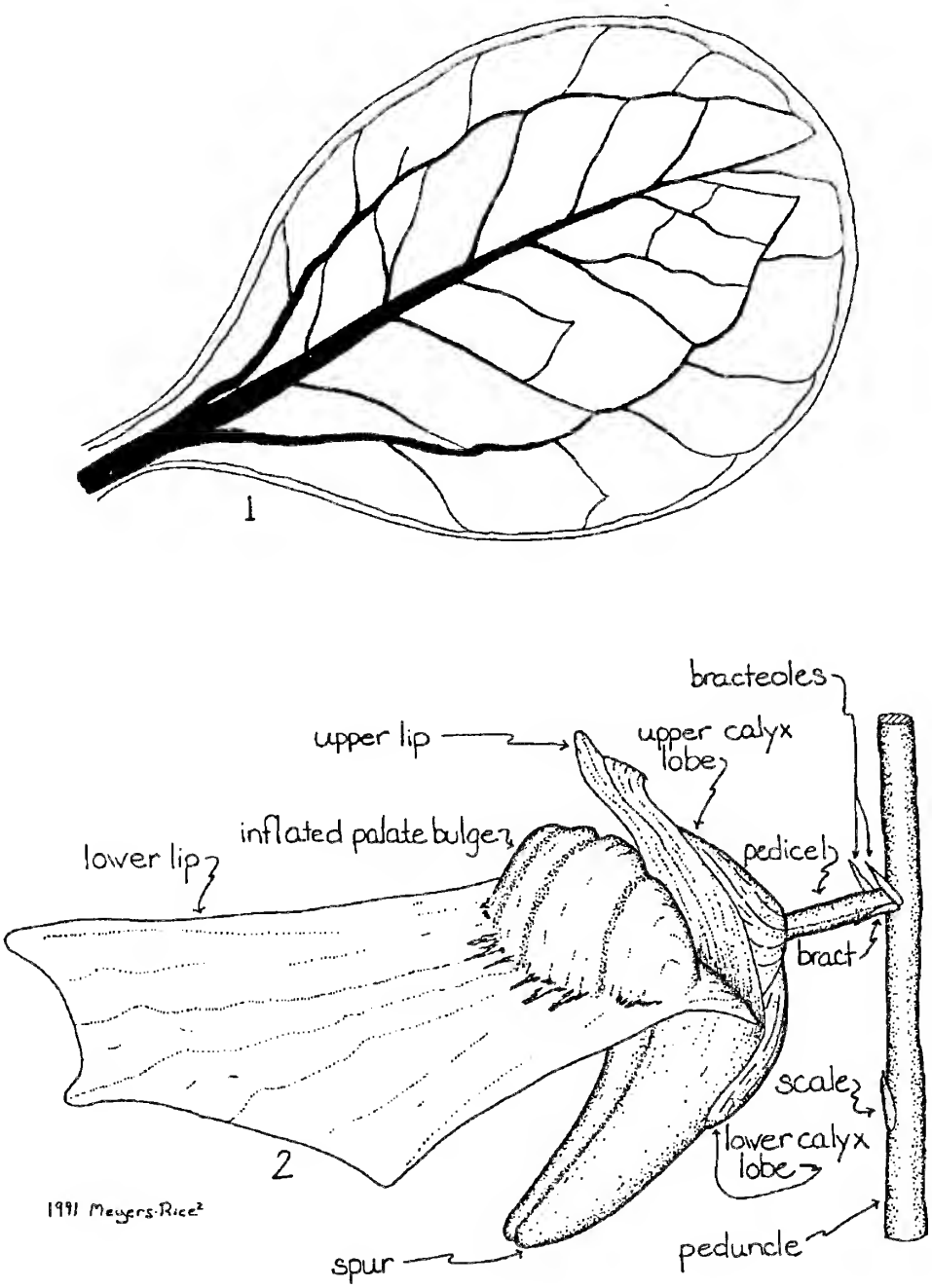

Figure 1: 1, a single U. *calycifida* leaf showing the complex network of veins; 2, a much magnified fictional *Utricularia* flower with relevant structures labelled.

In good conditions the plant will produce a thick vertical flower stalk (peduncle) that is round in cross section near its base. Often when *U. calycifida* produces a flower stalk, a second inflorescence will emerge near the first in about a week. The upper portion of the peduncle has subtle grooves and ridges along its length, producing an almost polygonal cross section. At maturity it can be more than 20 cm tall and have produced more than 20 flowers. The flowers are on short (up to 5 mm) horizontal or ascending pedicels. In Figure 1 I have drawn and labelled the parts of an archetypal flower to clarify my use of botanical terminology. Where each pedicel branches from the peduncle are three little (1 to 3 mm long) prongs—one bract and two bracteoles. These are very noticeable on this species. As in all *Utricularia* (except for those once classed as genus *Polypompholyx),* the calyx is divided into two sepals, or calyx lobes. Looking very closely (you may need a microscope) you'll notice the lower calyx lobe is forked at its tip and the sepal margins are minutely fringed. The name *calycifida* means "split calyx." Altogether, the pointed bracts, bracteoles, and fringed calyx lobes remind me of some sort of warrior or armoured beastie of Arthurian legend. Each combat equipped flower glares out over the field of battle-Sphagnum! Just a few days after each flower matures, it drops off and the wide sepals clasp together to protect the developing fruit.

The features described are common to all the clones of *calycifida* I have seen. The flowers, however, can vary greatly in color, size, and shape. None are scented, but this might occur in special or ideal conditions. In general, a *Utricularia* corolla can be divided into three main components: the upper lip, the lower lip, and the spur. In one of my forms of *calycifida* the 5—6 mm long upper lip arches close against the lower lip. It is only 3—4 mm wide where it emerges from beneath the upper calyx lobe, narrows smoothly to 1.5—2 mm in width, and then abruptly truncates in a rounded end. The lower lip is longer than the upper, 6—7 mm long. It is only 3—4 mm wide when it emerges from the calyx lobes, flares to 4—5 mm wide at mid-length, and then tapers to a point. The edges of the lower lip are often strongly reflexed or undulate. Like the upper lip it is pale lilac or white. The keel-shaped inflated palate bulge is longer than it is wide, and at its top, nearly hidden by the upper lip, is an orange-yellow splotch, margined with purple-brown. Much of the rest of the lower lip is speckled with small (1/4 mm diameter) purplish-brown spots. These spots also occur on the upper lip, but are stretched out into elongated streaks. The spur, usually paler than the lips, curves up to almost touch the lower lip at its tip. I call this form *calycifida* 'spotted flower' (Figure 2). Keep in mind this is not a registered cultivar name, it is just how I informally refer to the plant. The veins on this plant's leaves are purple only while the leaves are growing, and even then the coloration is mild—often purple is completely lacking in the leaves. This plant self pollinates readily, and if you let it will infest all of your other pots with viable seeds!

The second form of *calycifida* I grow is cultivated by many CPers in the U.S. Those that don't know its specific name call it *U.* sp. Venezuela. Even when it is not in flower it is distinguishable from 'spotted flower' because of differences in its foliage—it produces far fewer but larger leaves. Also, the veins on this form are bold and purple for the leaf's entire life span. I refer to this plant as *calycifida* 'purple veins' (another informal, unregistered name). The inflorescence of the two forms are similar except in the details of the corolla. In 'purple veins,' the corolla's upper lip is more erect and is only occasionally horizontal, and both the upper lip and spur are larger—up to 10 mm long each. But the chief difference between the flowers of the two forms is in the lower lip. Up to 1 cm long, the lower lip of 'purple veins' is slightly or clearly three lobed, not very undulate, and is much wider than in 'spotted flower.' The inflated palate bulge is approximately hemispherical. The coloration of this form is also different—the entire flower is lavender, except for a yellow blotch ringed with a very narrow white zone, located at the crest of the inflated palate bulge (Figure 3).

Figure 2: A flower of *U. calycifida* 'spotted flower.'

Figure 3: A flower of *U. calycifida* 'purple veins.'

Altogether, the flowers of *calycifida* 'purple veins' are the larger and more striking of the two. With excessive temperature, as often found in terrarium culture, the flowers tend to pale somewhat, especially as the inflorescence reaches the light fixtures. This plant produces seeds less commonly than does 'spotted flower,' but I've obtained seed by selfing the flowers.

Cultivation of *calycifida* is easy. It enjoys the conditions many tropical CPs like, and is an ideal terrarium subject. It is not at all picky about temperatures, provided you keep it warm, around 60—90°F, and in high humidity. For soil, I use dead or live *Sphagnum*—it is a large species so it usually does not become overwhelmed by live moss as long as you have fulfilled its other requirements. If I keep my plants too cool they slow in growth but don't die. I doubt they could survive a frost well. If you grow them too warm the flowers are often short lived, pale, and stunted. The plant's strongest dislike is bright sunlight—with too much sun it will only produce small

leaves that easily burn. Aphids and scale can be problems, and the best policy is prevention. If you have an infected plant and can't get rid of the pests by picking them off it might be best to find a subterranean, uninfested portion of your plant and start up a new pot.

In the wild the plant is found in shaded wet areas in northern parts of South America, i.e. Venezuela, Guyana, Surinam, and northern Brazil. With the recent surge in interest for the flora of these countries, it is likely new clones of the plant will be introduced to cultivation. I have already heard of several South American plants new to cultivation I suspect are *calycifida.* These new clones may have flowers with different colors and morphology from the ones I've described here, but the other characteristics (i.e. leaves, peduncle, bracts, sepals) are likely to be the same. The dimensions I have quoted in this article, especially of the flowers, should be viewed as variable. Under different conditions from mine, the flowers and leaves may be larger or smaller.

I'm beginning to experiment with selfing and crossing among the *calycifida* forms I have to see if the plants will grow true from seed. These experiments are incomplete but I'll publish them in a future issue of CPN as a letter to the editors. If you grow different forms of *calycifida* side by side, you may be surprised or skeptical that they are the same species—I know I was. However, Peter Taylor himself examined specimens I sent him and verified they were *calycifida.* I hope botanists will study the populations of these plants in the wild—perhaps some day the species will be split into two or more species or subspecies. Until then, they are all considered forms of one interesting polymorphic species.

I thank Don Schnell and Peter Taylor for comments and criticisms useful in the preparation of this article. I also thank Peter Taylor for examining my specimens of *Utricularia.*

Mechanisms Of Trap Movement 1: Rapid Growth in *Drosera, Dionaea* and Scientific Notions Of How Venus's Flytraps Close[1]

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Plant and fungal movements often result from differential curvatures or the relative change in size of the cells on each side of a structure. When the term differential curvature is used it usually brings to mind processes in which different relative growth rates on each side of a structure result in its bending. However, it can be seen in figs. 1-3 that differential growth is only one mechanism by which such changes could occur. Others involve a relative loss or gain of turgor of the cells on each side of the structure.

There are three genera of the family *Droseraceae* with movements involved in their trapping mechanisms. In each of these genera it has long been clear that the movements were the result of differential curvatures. By 1934 major papers on the mechanisms of movements in each of the three genera proposed different mechanisms by which these curvatures would be brought about. *Drosera* tentacles were thought to move by a growth mechanism involving a relatively rapid increase in wall plasticity on the abaxial (back) of the tentacles (Hooker, 1916), *Dionaea* traps were thought to move by a rapid gain in turgor by the abaxial (back or outer) side of the trap lobes[2] (Brown, 1916). *Aldrovanda* trap lobes were thought to move by a rapid loss of turgor in the abaxial (inner) side of the trap lobes (Ashida 1934; Figs. 1-3).

Of course by 1979 any well trained plant physiologist believed that slow plant movements, such as those of phototropism, were growth movements and rapid nastic movements were all due to the rapid loss of turgor. Certainly the mechanism of *Mimosa* was well known (Sibaoka, 1980) and we assumed it could be generalized to all rapid movements in plants. This assumption was not just implicit it was often openly stated. Most of us just assumed that *Dionaea* moved by a turgor mechanism. Few read 1916 papers on *Dionaea* and those who did thought that Brown had to be mistaken since a rapid turgor gain seemed unlikely. His data, showing an irreversible increase in cell size of the outer lobes of the trap on closure, which he referred to as "growth" even though he invoked a turgor mechanism to accomplish it, had to be in error since it did not fit with our preconceptions. Ashida's (1934) model for *Aldrovanda* was much more in keeping with our ideas and so I considered that to be the mechanism of *Dionaea* trap lobes as well (Williams,1973; Williams,1976). I certainly received no arguments from my colleagues for such orthodox ideas however inconsistent with the data they might be.

Barbara Pickard and I, when writing a review paper for a July 1979 Symposium in Madison, WI on Plant Movements (Williams and Pickard, 1980)[3], decided to present the paper as a comparison of the *Drosera* mechanisms with *Dionaea* mechanisms since it had become apparent that there were many parallels in these two genera (Williams, 1976). When the data was lined up side by side it became apparent that all evidence for both plants was parallel. Despite this, irreversible changes in cell length in *Drosera* tentacles were said to be due to growth by Hooker (1916) and irreversible changes in cell volume in *Dionaea* were ascribed to a mechanism involving a rapid turgor gain by

Brown (1916). Brown's hypothesis lacked appeal because a rapid gain in turgor seemed unlikely and because it was clear that the trap opened by a growth mechanism and got successively larger each time it moved. Robert Clealand had introduced the concept of acid growth (Clealand, 1980)[4], a process which could be quite rapid, and Barbara Pickard and I (Williams and Pickard, 1972) had demonstrated that the "slow" growth response of *Drosera* tentacles could occur within 10 to 15 sec. It was not unthinkable that the rapid response of the *Dionaea* trap was due to acid growth.

Alan Bennett (then a graduate student at Cornell, now a professor at the University of California at Davis) and I began a series of experiments in Roger Spanswick's laboratory at Cornell and at Lebanon Valley College in Pennsylvania which confirmed the accuracy of Brown's measurements. These also demonstrated that the trap would not close if wall acidification was prevented by neutral buffers and that trap closure would occur if wall acidification was artificially caused with acid buffers (Williams and Bennett, 1982). The only thing that bothered us was that the gradient of hydrogen ions and electric potential in plant cells is such that hydrogen ions should passively flow into the cell instead of out!

It was at this point I remembered Mark Jaffe's (1973) experiments on ATP changes in the midrib during trap closure. This paper, which was my inspiration when I wrote *Why a flytrap is not a Bear Trap* (Williams, 1973), had never made sense to me. The midrib is not where the movement takes place, the units used in the paper did not make sense and I, still believing in Ashida's mechanism, saw no logical reason to look for ATP changes. Suddenly there was a reason to expect ATP changes. Maybe every cell in the excitable zone of the trap, or at least the trap epidermis, responded in the same way. Maybe Jaffe's ratios would hold even if his units did not make sense. Alan Bennett and I froze trap lobes in the open and closed condition and found that Jaffe's ratios did hold and that about 30% of the trap ATP disappeared during closure (Williams and Bennett, 1982; Fig. 3). When we spoke to Roger Spanswick, who was on sabbatical in California, on the phone that evening Alan and I were saying we had a "proton cannon" (the mechanism that uses ATP to move hydrogen ions across cell membranes is often called a "proton pump"). It appeared the power to quickly move hydrogen ions across the membrane was there.

Within the Droseraceae *Dionaea* and *Drosera* have the same mechanism causing their rapid, action potential initiated movements. Although taxonomically it might have been expected and evidence had been sitting around since 1916 it still came as something of a surprise. *Aldrovanda* the other genus in Droseraceae with an active trap is still reported to move by Ashida's rapid turgor loss mechanism (Ashida, 1934; Iijima and Sibaoka, 1983). This may be so but the evidence is not yet firm. If the *Dionaea* and *Aldrovanda* mechanisms differ we are in for another surprise.

Literature Cited

Ashida, J. (1934), Mem. Coll. Sci. Univ. Kyoto, Ser. B 11: 55-113.

Brown, W. (1916) Am. J. Bot. 3: 68-90. Clealand, R. (1980) In Plant Growth Substances 1979. F. Skoog, Ed. (Springer-Verlag, Berlin), pp. 71-78.

Hooker, H. (1916) Bull. Torrey Bot. Club 43: 1-27

Iijima, T. and T. Sibaoka, (1983) Plant & Cell Physiol. 24: 51-60.

Sibaoka, T. (1980) In Plant Growth Substances 1979. F. Skoog, Ed. (Springer-Verlag, Berlin), pp. 462-469.

Williams, S. (1973) **The Carnivorous** Plant Newsletter. 3: 23-24.

Williams, S. and A. Bennett (1982) Science 218: 1120-1122.

Williams, S. and B. Pickard (1972) Planta 103: 222-240.

Williams, S. and B. Pickard (1980) In Plant Growth Substances 1979. F. Skoog, Ed. (Springer-Verlag, Berlin), pp.470-480.

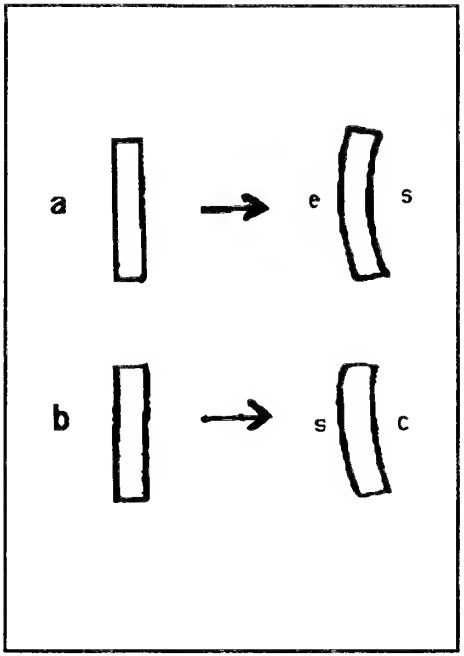

Figure 1

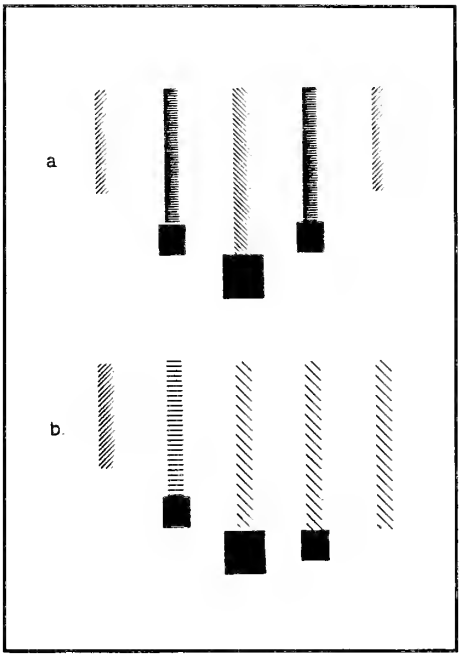

Figure 2

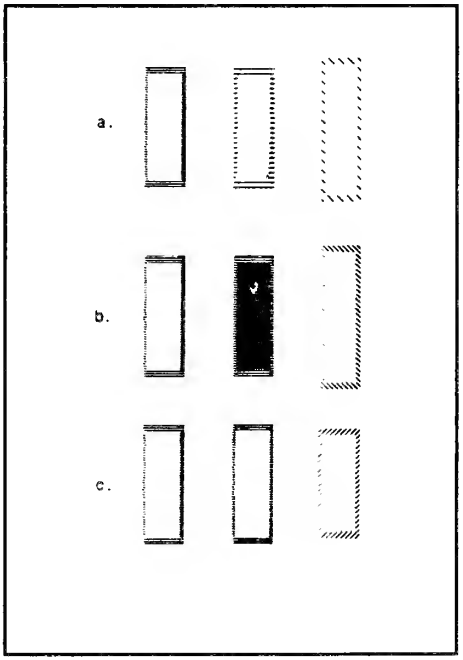

Figure 3

Figures

Figure 1. Movements of plant structures often occur as a result of curvatures caused by differential explanation and/or contraction on the two sides of the structure. **a.** Differential explanation (in this instance explanation on side "E" with no change on side "S") results in the curvature illustrated. **b.** Differential contraction (in this instance contraction on side "C" with no change on side "S") results in the curvature illustrated.

Figure 2. Elastic structures, such as rubber bands, return to their original shape when tension on them is relaxed. Plastic structures, such as modeling clay, remain distorted after tension on them is released. Many things, such as a child's balloon, have some of each property and do not totally return to their original shape when first stretched then allowed to spring back. **a.** A perfectly elastic cell wall being stretched by a weight and returning exactly to its former state just as a perfect spring would. **b.** A perfectly plastic cell wall which remains completely distorted when stretched by a weight. Real cell walls would vary between these two extremes. The early phases of plant growth have long been known to involve an increase in plasticity (decrease in elasticity) of the cell walls. Hooker (1916) was expressing prevailing ideas about plant growth when he invoked this mechanism to account for the growth of *Drosera* tentacles.

Figure 3. a. Cell expansion can result from a loosening and an increase in the plasticity of cell walls that allows them to be extended by the pressure that is in plant cells (turgor pressure). Even though the turgor pressure is necessary for such an expansion it is the change in the wall properties that allows it to occur. Such an event is involved in the early phases of growth and movements caused by such a mechanism would be called a growth movement. **b.** Expansion can also occur by the cell producing or taking up additional dissolved substances such as sugars or potassium ions which increase the osmotic pressure and draw water into the cell resulting in an increase in turgor pressure. Here again it is the turgor pressure that expands the cells but this time the movement is initiated by an increase in the concentration of dissolved substance and the walls merely act as springs being stretched by the pressure created by the water that is taken up. Movements caused by such a change would be called turgor movements. **c.** Reduction of the size of the cell can also occur by a turgor change. Loss of dissolved sugars or ions from the cell would decrease the osmotic pressure and result in the loss of water from the cell. As a result the cell would shrink in size. Here again the cell wall need only act in an elastic way as a spring. Thus turgor movements can occur due either to expansion from a turgor gain or shrinkage from a turgor loss. Combinations of the above mechanisms are also possible although more complex.

Footnotes

[1] This is part one of a projected three article series by this author on the subject.

[2] Brown (1916) referred to these movements as growth movements, probably because he had determined that they involved an irreversible enlargement of cells, but his hypothesized mechanism for the movements was a turgor gain that caused a plastic expansion of the walls of the outer epidermis during the movement.

[3] The symposium was attended by Takao Sibaoka who gave the paper before ours in the session. He reported the first intracellular recordings of action potentials in *Aldrovanda* (1980). I had a pleasant conversation with Dr. Sibaoka afterwards at the banquet.

[4] At the same conference in another session Cleland (1980) presented a review of his then relatively new theory on "acid growth" as the mechanism of auxin action. He may have attended our session.

Growth Effects of Mineral Nutrients Applied to the Substrate or onto the Leaves in Four Carnivorous Plant Species

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The majority of terrestrial carnivorous plants (CPs) grow on mineral poor and wet soils. Moreover, another common characteristic of carnivorous plants is the weakly developed root system. The limited capacity for mineral nutrient uptake by roots of CPs may be partially compensated by the absorption of nutrients from the caught prey. The question of the relative contributions of roots and trapping leaves to the nutrient supply necessary to normal growth has long been considered. Now, it is accepted that no CP can sustain its growth when fed only with insects as prey via leaves (Juniper et al., 1989, p. 130). However, normal growth and development of CPs can take place even when no feeding with insects is applied. Juniper et. al. (1989, p. 130-134) reviewed in their recent monograph the results concerning the growth effects of mineral nutrients added to root substrate (solution) or feeding with small animals on the leaves of CPs. Feeding with insects on the leaves always led to an appreciable enhancement of growth in greenhouse cultivation in various species of CPs grown in mineral poor media. This finding has recently been confirmed also in natural populations of *Drosera intermedia* and *D. rotundifolia* (Thum, 1988).

Feeding the CPs with insect bodies is not the only way of feeding and it may be successfully replaced by adding small pieces of agar gel impregnated with nutrient solution on the trapping parts of leaves (Karlsson & Carlsson, 1984). The authors found that it was mainly phosphate added as agar gel droplets on the leaves that enhanced appreciable growth in the common butterwort, *Pinguicula vulgaris*, and led to an increase of total amount of both phosphorus and nitrogen in plants. It is remarkable that the increase of the total phosphorus absorbed by the P-fertilized plants, as compared with controls, was about 1.5-2.5 times higher than the dose added on the leaves. The same phenomenon was observed also for nitrogen in the same species when the plants were fed with small flies (ratio of 1.6; Aldenius et al., 1983). These results suggest that the leaf uptake of phosphate (and perhaps also of other nutrients) stimulates the effective uptake of many mineral nutrients by roots.

Many CPs are able to enhance markedly their growth and nutrient content in tissues after mineral nutrients have been added to a nutrient-poor substrate (or solution; for review see Juniper et al., 1989, p. 130-134). However, negative interactions between feeding with insects on leaves and nutrient uptake by roots also takes place in CPs and they reduce the growth and total nutrient content in plants (Chandler & Anderson, 1976). In any case, the uptake capacity of roots for mineral nutrients from soil is relatively low in CPs. Moreover, when the roots of CPs were grown in nutrient-rich soils, growth was significantly reduced and the plants lost some features of carnivory (Roberts & Oosting, 1958; Eleuterius & Jones, 1969). Thus, mineral nutrients may be added only with caution to the substrates for cultivation of CPs. The

above fact that the growth of CPs may be considerably enhanced by moderate mineral fertilization both of substrates and on trapping leaves might have a practical application in the cultivation of CPs. In this paper, we compare the growth effects of mineral nutrients added either to a peaty substrate or on trapping parts of leaves as well as the effect of substrate alkalization in four species of CPs. The effectiveness of utilization of added nutrients in plants is discussed.

Four species of CPs were used for experimental cultivation: *Drosera adelae* F. Muell., *D. aliciae* Hamet, *D. capillaris* Poir., and *Dionaea muscipula* Ell. These species originate from various continents. They are often cultivated by growers. An acid fen soil (l kg of fresh weight, FW) with washed sand (0.5 l) was used as a substrate in all experimental variants. The fen soil was obtained from an acid fen near Trebon town (Trebon Biosphere Reserve, South Bohemia, Czechoslovakia). It originated from the litter of sedges and common reed overgrown by a wet needle-leaf forest. It was of brown-black color, with short filaments.

Some basic analyses of the fen soil were performed. Data are available to show that the content of accessible (exchangeable) Ca, Mg, Na, K, and Fe in the fen soil was sufficient to support plant growth. Dry weight, DW, (105 °C for 3 hours) amounted to 24.9% of FW. One gram of the fen soil (FW) was shaken with 5 ml of distilled water; the pH was in average 3.97, rather low for a fen soil, and it dropped to 3.35 after adding KCl (exchange pH value) indicating a high cation-exchange capacity. Electrical conductivity of the soil suspension was rather low (47 $\mu S.cm^{-1}$) indicating that the fen soil was nutrient-poor. Thus, the fen soil pH was about 4.0 in all but one variant.

In one variant.the fen soil was alkalized by means of $NaHC0_3$ to a pH of about 5.2 prior to cultivation. Titration of the fen soil suspension with 0.1 $mol.l^{-1}$ $NaHC0_3$ revealed that its buffering (and/or neutralization) capacity was very high. To reach pH 5.2, it was necessary to add 21.5 mmol (1.81 g) $NaHC0_3$ to 1 kg (FW) of the fen soil (i.e., 86 mmol per kg of DW. We also estimated how easily the acid fen soil could be neutralized by $HC0_3^-$ from the tap water which was used for watering in all variants. We found that, owing to the high buffering capacity of the soil, as much as 50 ml of tap water might be added to 1 g of the fen soil (FW) for pH to rise by only 1.0 unit (from 4.0 to 5.0). The $HC0_3^-$ concentration of the tap water used was about 0.7 $mmol.l^{-1}$ (42 $mg.l^{-1}$) its pH 7.2-7.5, and electrical conductivity about 76 $\mu S.cm^{-1}$. The mean $N0_3^-$ concentration in the tap water was about 3 $mg.l^{-1}$ (i.e., about 0.7 $mg.l^{-1}$ of N03-N); other N salts were not present. Phosphate concentration was extremely low, within 3-5 $\mu g.l^{-1}$ of $HP0_4$-P. The mean Ca^{2+} concentration was about 25 $mg.l^{-1}$. K^+, Na+, Mg2+, and $S0_4^{2-}$ ions were also present in low concentrations.

On 16 February 1990, ten small plants of each of the four species were planted together on the fen soil with sand (see above) in each of four glass aquaria (18 x 20 x 20 cm); 0.25 kg DW of the fen soil was present in all of them. The substrate depth was about 5.5 cm. Uniform seedlings of *D. aliciae* and *D. capillaris*, plantlets of *Dionaea* from meristem tissue culture, and *D. adelae* plantlets arising from root regeneration buds were used. The aquaria with plants, covered by pieces of glass, were placed on a window ledge of SW orientation. Tap water was added to keep the water table at about a half of the substrate depth. Over the whole cultivation period, 2.5-3.0 l of tap water was added to each aquarium. The temperature inside the aquaria ranged from 18 to 27 °C. In summer, the aquaria were shaded by thin paper sheets preventing them from overheating. the position of individual aquaria on the window ledge was rotated regularly at one-week intervals.

Variants of mineral nutrition

Each aquarium with 10 plants of each of the four CP species represented one variant. The variants were as follows:

1. Control, denoted as "C". The substrate was watered with tap water only.
2. Variant whose substrate was alkalized to pH of about 5.2 (see above) by adding $NaHCO_3$ prior to experiment. It is denoted as "ALK". The substrate pH dropped during the experiment and was reset at pH 5.2 by further additions of $NaHCO_3$.
3. Variant (denoted as "NS-S") whose substrate was homogeneously fertilized by 50 ml of mineral nutrient solution of the following composition: (in mg.l-1): NH_4NO_3 200.1, KH_2PO_4 100.7, $MgSO_4$·7H2O 101.1, $CaCl_2$ 110.0, $FeCl_3$ 2.6. Thus, the mean nutrient concentration in the substrate rose by (in mg of element·kg^{-1} DW): N 14.0; P 4.6;, K 5.8; S 2.6; Ca 8.0; Mg 2.0; Cl 17.6; Fe 0.18.
4. Variant (denoted as "NS-L") where the same mineral nutrient solution as in NS-S was dropped on trapping parts of the leaves once a week. A droplet of 1 or 2 µl was applied on either one or two of the leaves of each plant. Thus, the total volume of the nutrient solution per plant amounted to 92 µl in *D. adelae*, 38 µl in *D. aliciae*, 92 µl in *D. capillaris*, and 46 µl in *Dionaea* during the whole cultivation.

On 3 March 1990, 15 days after planting, the length of the longest leaf was measured in all plants. This date is considered to be the onset of proper cultivation as plants could be injured during planting. The leaf length was measured also on 16 May, 20 June, and 6 October. The cultivation was finished on 6 October, 217 days after the date of the first leaf measurement. The length of the longest root was measured in all plants. Both roots and shoots of 10 plants of each variant were dried (105 C for 3 hrs) and weighed. The dead biomass was not separated. It was negligible in all *Drosera* plants but it could amount to some 30% of the *Dionaea* shoot weight and even 40-50% in the NS-L variant. The leaf and root lengths were always expressed as mean value ±2.SEM.

The results (Figs. 1-4; Table I) show that all control plants (C) grew rather slowly. The length of the longest leaf or root may be understood only as a rough measure since it did not reflect the total plant biomass too closely and, moreover, it had a high variance. Except for *D. capillaris*, no distinct increase of plant biomass took place in the alkalized substrate (ALK). In all *Drosera* species, when supplied with mineral nutrients either to the substrate (NS-S) or on the leaves (NS-L), their shoot and root growth was markedly promoted and their total biomass rose to about 2.4 to 17 times that in controls. The vigorous growth of *Drosera* shoots more closely correlated with root growth although the shoot/root ratio (dry weight) was appreciably higher than in controls only in NS-L. *Dionaea,* however, did not react positively to nutrient supply applied either to roots or on leaves. In the latter case, the extremely high ratio of dead biomass was probably due to frequent closing of the leaf traps.

A remarkable phenomenon was observed in all *Drosera* species in the NS-L variant. The amount of mineral nutrients in the total plant biomass minus the nutrient amount in the controls was many times higher than that added on the leaves as nutrient solution during the whole experiment (Table II). Published data on the mean nutrient content in dry biomass of *Drosera* were drawn from Juniper et al. (1989; Tab. 11.1, p. 230) and Dykyjová & Drbal (1984; Tab. 3, p. 82). The following values were used (in mg of element.g^{-1} DW): N 11; P 1.25; K 11; Ca l; Mg 1.95; Fe 0.5. Similarly, the increase of total plant biomass in the NS-S variant theoretically was higher (against the controls) than would be expected from the nutrient supply to substrate (for N about 1.4 times~ K 3.4, and Mg 1.8 times).

The present results support fully the literature data in that the growth of CPs can be promoted appreciably by mineral nutrient supply either to the substrate or directly on the leaves (cf. Juniper et al., 1989, p. 130-134). This finding is of a very practical importance for all growers of CPs as it is very easy and cheap to add a mineral nutrient solution to substrate or spray the leaves of CPs with the same solution. The sprayed nutrients that fall on the substrate can be taken up by roots. The greatest advantage

Table I. Results of growth experiment after 217 days of cultivation. Variants: C, controls; ALK, substrate alkalization; NS-S, nutrient solution added to substrate, NS-L, nutrient solution dropped on leaves. Length of the longest root was measured in 10 plants. DW, dry weight.

Variants	Root length ±2.SEM (cm)	Shoot DW (mg)	Root DW	Shoot DW / Root DW
A. Drosera Adelae				
C	4.1 ± 1.2	15.1	4.5	3.3
ALK	3.0 ± 2.3	7.7	2.9	2.7
NS-S	10.6 ± 3.2	43.0	13.0	3.3
NS-L	8.1 ± 1.9	38.4	8.9	4.3
B. Drosera aliciae				
C	0.52 ± 0.11	0.24	0.04	6.0
ALK	0.65 ± 0.20	0.31	0.06	5.2
NS-S	3.6 ± 0.6	4.3	0.62	6.9
NS-L	1.3 ± 0.6	3.1	0.14	21.9
C. Drosera capillaris				
C	2.7 ± 1.1	4.7	0.70	6.7
ALK	2.3 ± 0.5	8.3	0.76	11.0
NS-S	7.3 ± 2.6	15.3	2.3	6.6
NS-L	6.5 ± 2.7	19.9	2.4	8.4
D. Dionaea muscipula				
C	3.0 ± 1.3	27.5	0.96	28.7
ALK	2.0 ± 0.7	14.0	0.65	21.5
NS-S	3.2 ± 1.0	19.7	0.63	31.3
NS-L	3.5 ± 1.5	28.3	0.89	31.8

Table II. The theoretical increase of the total nutrient content in the NS-L variant as compared with controls divided by the total nutrient content dropped on the leaves (i.e., the effectiveness o utilization of the nutrient added).

Species	N	P	K	Ca	Mg	Fe
D. adelae	47.2	16.4	114.	7.5	58.8	169
D. aliciae	12.1	4.2	29.2	1.9	15.0	43.2
D. capillaris	28.9	10.1	70.0	4.6	36.0	103

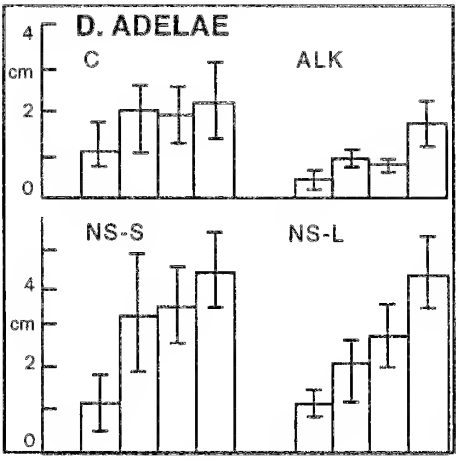


Figure 1

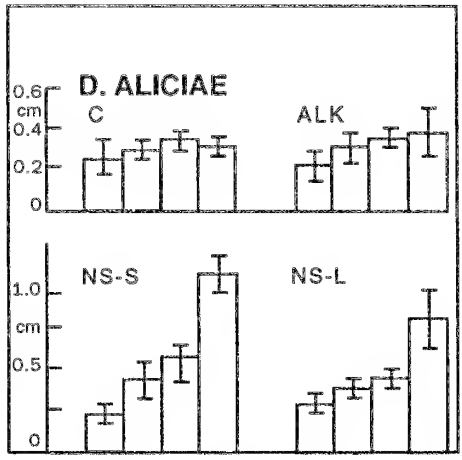


Figure 2

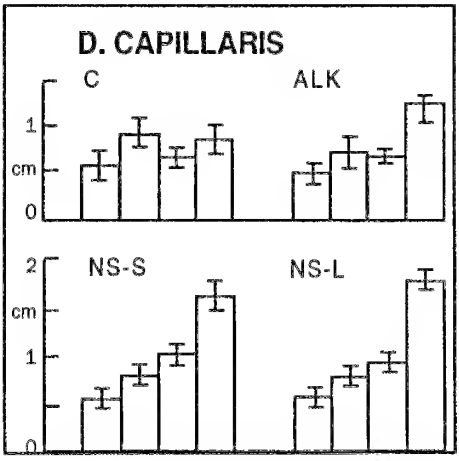


Figure 3

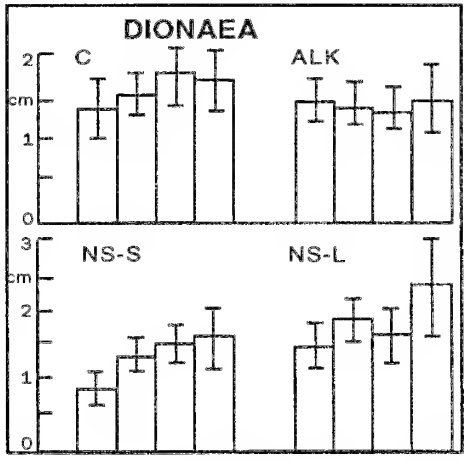


Figure 4

Legends to the figures:

Figure 1. The mean length of the longest leaf in *D. adelae* in the course of 217-day cultivation ±2.SEM; n=10. Variants: C=controls, ALK=substrate alkalization, NS—S=nutrient solution to the substrate, NS-L=nutrient solution dropped on the leaves. Date of measurements from left to right in tetrad of columns: 3 MAR, 16 MAY, 20 JUN, 6 OCT.

Figure 2. The mean length of the longest leaf in *D. aliciae*. For explanation see Figure 1.

Figure 3. The mean length of the longest leaf in *D. capillaris*. For explanation, see Figure 1.

Figure 4. The mean length of the longest leaf in *Dionaea muscipula*. For explanation, see Figure 1.

of a mineral nutrient solution is that it cannot get mouldy. However~ it is necessary to take into account that overfertilization of a substrate can lead to reduction of growth (Roberts & Oosting, 1958; Eleuterius & Jones, 1969). There was a relatively low growth rate in all plants during our experiment, probably due to low light intensity. Thus, it is reasonable to suppose that if the external growth conditions had been optimal, the growth effects of added nutrients would have been even more distinct. *Dionaea* however, seems to be adapted to a very low nutrient content in the soil and also its capacity to absorb mineral nutrients by leaves is negligible.

The aim of substrate alkalization was to find out whether the growth of CPs might be promoted by a pH higher than that in a natural acid fen soil. Except for *D. capillaris*, ecologically a very resistant species, alkalization resulted in the same or reduced growth (Figs. 1-4; Table I). Similarly, Rychnovská-Soudková (1953, 1954) found the optimal growth of *Drosera rotundifolia* to be in a diluted nutrient solution at pH as low as 3.0. The very low pH protected the plants from a high Ca^{2+} concentration in the solution and NO_3^- was used efficiently as a source of N. At pH above 5.0 the growth was promoted significantly by NH_4^+ whereas Ca^{2+} and NO_3^- inhibited it. In general, it may be concluded that *Drosera* plants are more susceptible to elevated concentration of nutrients at higher pH values.

As Table II shows, the growth of the three *Drosera* species in the NS-L variant was so vigorous, compared to the controls, that the amount of major nutrients theoretically accumulated in the biomass was many times higher (for N, P, K, 4 to 114 times) than could be absorbed from the nutrient solution dropped on the leaves. However, the numerical data in Table II represent only rough estimates (approximate values) of the true ones as literature data on the nutrient content in *Drosera* species were used instead of analyses. Except for iron, the content of other elements was not too variable. Thus, it is reasonable to assume that the true nutrient content in biomass did not differ from that used for calculation more than by ±50%. i.e., the data in Table II lie also within this range. The nutrient content per biomass unit of CPs which were supplied with mineral nutrients or insects was not too different from that of controls and the former was usually somewhat higher (Chandler & Anderson, 1976; Aldenius et al., 1983; Karlsson & Carlsson, 1984). Therefore, the data shown in Table II could represent conservative estimates.

How can this intriguing phenomenon be explained? It is quite clear that the CPs in the NS-L variant absorbed the whole remaining amount of nutrients by roots from the fen substrate. Though chemical analysis of the fen soil used is lacking, it may be deduced from analyses of other similar fen soils of the Trebon basin (Dykyjová & Drbal, 1984) that the content of all nutrients in the fen soil used was high enough to ensure the observed increase of biomass. Moreover, small amounts of nutrients were introduced into the substrate in the tap water. Now, the question arises how dropping small amounts of nutrients on the leaves could cause the roots in the NS-L variant to take up so much nutrient from the relatively poor substrate? Two independent mechanisms may be suggested; they might also operate simultaneously. a) The supply of nutrients on the leaves of relatively small plants in the early phases of experiment could lead to a partial promotion of root growth. The longer roots would be capable of absorbing more nutrients from the poor substrate thus supporting more shoot growth. In the course of time, this positive feed-back mechanism could produce roots which were about 2-2.5 times longer than those of controls (Table I). Considering the three-dimensional distribution of these roots in the substrate, they could theoretically come in contact with about 8-15 times greater substrate volume than the controls. Probably, it could not be sufficient (cf. Table II). b) As stated by Aldenius et al. (1983) and Karlsson & Carlsson (1984) who also found this phenomenon for N and P, the leaf nutrient supply might result in an increased efficiency of nutrient uptake by roots from the substrate.

The latter authors concluded that the main candidate for the stimulation of nutrient uptake by roots of CPs was probably phosphate that had been absorbed by leaves. This conclusion was based mainly on the fact that the leaf uptake of phosphate itself led to a significant increase in the total N content of the plants. However, the leaf uptake of N and microelements interacted positively with that of P indicating that the stimulation of nutrient uptake by roots was more complex.

The same explanation may hold also for the NS-S variant. As all CPs in our experiment were grown in very humid air, the effect of dropping the nutrient solution on the leaves cannot be explained in terms of an increase in the supply of water itself.

Acknowledgment

The authors are indebted to Dr. Miloslav Studnicka, Head of the Botanical Garden in Liberec, Czechoslovakia, for his providing the experimental plants and valuable comments. Sincere thanks are due to Dr. Laurie E. Friday, University of Cambridge, U.K., for language correction of the manuscript and for helpful criticism.

References

Aldenius J., Carlsson B., Earlsson S., 1983. Effects of insect trapping on growth and nutrient content of *Pinguicula vulgaris* L. in relation to the nutrient content of the substrate. New Phytol. 93: 53-59.

Chandler G.E., Anderson J.W., 1976. Studies on the nutrition and growth of *Drosera* species with reference to the carnivorous habit. New Phytol. 76: 129-141.

Dykyjová D., Drbal K., 1984. (Chemical composition of peat bog and acid fen vegetation in the Trebon basin.) In Czech. Preslia (Prague) 56: 73-91.

Eleuterius L.N., Jones S.B., 1969. A floristic and ecological study of pitcher plant bogs in south Mississippi. Rhodora 71: 29-34.

Juniper B.R., Robins R.J., Joel D.M,, 1989. Carnivorous Plants. Pp. 353, Academic Press Ltd, London.

Karlsson P.S., Carlsson B., 1984. Why does *Pinguicula vulgaris* trap insects? New Phytol. 97: 25-30.

Roberts P.R., Oosting H.J., 1958. Responses of Venus fly trap (*Dionaea muscipula*) to factors involved in its endemism. Ecol. Monographs 28: 193-218.

Rychnovská-Soudková M., 1953. Study on mineral nutrition of *Drosera rotundifolia* L. I. Influence of calcium as an important physiological and ecological factor. In Czech. Preslia (Prague) 25: 51-66.

Rychnovská-Soudková M., 1954. Study on mineral nutrition of *Drosera rotundifolia* L. II. Root sorption of inorganic nitrogen. In Czech. Preslia (Prague) 26: 55-66.

Thum M., 1988. The significance of carnivory for the fitness of *Drosera* in its natural habitat. 1. The reactions of *Drosera intermedia* and *D. rotundifolia* to supplementary feeding. Oecologia 75: 472-480.

Want Ad

Thomas Johnson, P.O. Box 12281, Glendale, CA 91224. Phone (818) 248-1623. *Drosera schizandra, D. villosa,* and several Mexican Pinguiculas for trade.

The "Seasonal Bog" Garden
An Alternative Method of *Sarracenia* Cultivation for the Northern* Grower

By Jerry B. Stahle, Sr.
700 Mulberry St., York, PA 17403
Phone (717) 846-3635

When I stepped into my house just back from the garden center, with a smile on my face from ear to ear, holding out in front of me for my wife to see, a $5.29 pot of *S. purpurea*, I never imagined that my lifestyle would become intoxicated by the secretion of nectar.

I was always curious about nature. As a child, the excitement I had running up to my grandma's large concrete lily pond hoping to see strange creatures and later as a teenager, exploring the local fresh water marshes every Sunday while the family got together for dinner. I was always bringing some form of plant or animal life home to study or donate to my high school science classes. Wetland ecology and the natural world in general established a place in my heart during those formative years.

*The author lives just inside zone 6, with an average min. temp. of -10 - O° F. and classifies all N.A. CP growers living in zones 7 - 2 (regardless of latitude), as outlined on a **U.S.D.A. Hardiness Zone Map** as "northern".

After four years in the military during the Vietnam Conflict, I made it a point to give my family the opportunities to experience the wonders of pond life as I had years before.

But, I never had the chance to become acquainted with bogs; and the day my wife came home from the store with a jewellike sundew, I was instantly enchanted. The plant lived for my wife about two months. It was during this time that I told her that I decided to grow CP.

My first year was very challenging and rewarding. In addition to taking notes on all manifestations of my pot of *S. purpurea ssp.*_ due to cultivation, buying three *Dionaea* plants and the joy of locating my first colony of *D. rotundifolia* while doing survey work (Stahle 1985:52). I was trying to obtain books on CP wherever possible, but it was also a very disturbing and frustrating time. Feeling responsibility to my plants, I was afraid to turn my back on them for more than a day or two, which warned me of the pitfalls of overextending oneself beyond one's time and facilities. As a result, I sat down and made a critical evaluation of my long term interests, incorporating Koopowitz's school of thought (Koopowitz & Kaye, 1983). I must admit that I bit my lip hard a few times; but I decided to limit myself to N.A. CP.

I agree with the philosophy that CP are not house plants (Bell 1976:vi), and should be grown outdoors to achieve maximum growth potentials (Hummer 1979:78, Schnell 1976:96) as well as affording them the natural setting that their beauty deserves. But,in the back of my mind, I was constantly haunted by the fact that I lived in a northern environment. I had to establish a method of cultivation to accommodate southern *Sarracenia* or be content with growing a few selected plants (Hummer 1979:78, Schnell 1976:100,109, Slack 1980:192), which I wasn't.

Another thing that bothered me was the sparsity of people growing CP which I felt was due, in part, to lack of an easy, standardized method of cultivation and a more affordable set-up. Once I started appraising greenhouses and calculating costs and overhead, I was convinced that this system's costs prevented the northern market for CP from expanding, at least comparable to the water lily garden trade.

With these thoughts in mind, it became obvious that a standardized system of outdoor cultivation for a wider spectrum of growers was needed. Good cultivation, affordable set-up, plus minimum operating costs and maintenance became primary objectives.

A review of literature on outside cultivation of CP was made in order to learn about technologies in use, while culling those technologies that I felt could be incorporated into the projected system.

The following information is the result of the author's attempt to achieve the above goals, plus notes since 1986.

If one finds it heartbreaking or unacceptable to have heavy rain, hail, nibbling insects or animals take their toll of foliage (Hummer 1979:95, Schnell 1976:109, Slack 1980:193), then bog gardening may not be for you. I shook many a fist at robins which are my biggest aggravation. All things aside though, the rewards and gratification of outdoor cultivation to me outweigh any bird or starving insect.

The primary tools needed are: BASKETS. Swenson briefly mentioned: "We have found that plants placed pot and all into the soil in a bog or moist woodland location will thrive all summer". (Swenson 1977:10).

Realizing the significance of such flexibility, I crossed over to the water lily trade and adopted their planting baskets (Figure 1). Made of a top quality composition to withstand water and handling, these baskets allow water to pass through the compost in a more natural way. They also provide good aeration of compost, prevents salts from accumulating and buffers the pH of your potting compost through the natural process of seepage. Root systems are also free to expand at will, reducing rootbinding. With these factors recognized, repotting is less frequent, reducing root disturbance which *Sarracenia dis*like with a passion (Schnell 1977:162, 1978:10, 1980:111), at the same time making it possible to rearrange your bog to obtain different effects. At the onset of freezing weather, plants can then be "lifted" for storage.

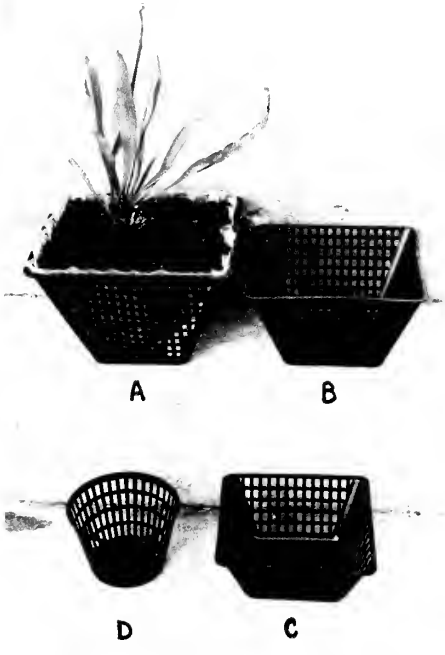


Figure 1. Planting Baskets

Sold in association with these planting baskets are burlap liners called Hessian Squares. These are used to prevent loss of soil. Feeling that the decomposition of such liners would add unknown trace elements to my *Sarracenia* compost and/or alter the pH, I substituted the burlap with the purchase of what is commonly called "bridal veil". This item is less bulky and very open in structure.

The baskets should also be modified to give better drainage for bog use by drilling nine 1/2 inch size holes through the bottom section in all square baskets regardless of size (not shown).

Figure 1 shows sizes available: A - Large square 10 inches by 7 inches deep; B - Medium square 9 inches by 5 1/2 inches deep; C - Small square 7 1/2 inches by 3 1/2 inches deep; D - Round 5 3/4 inches by 4 inches deep. All measurements are inside. I strongly suggest the use of baskets that are not less than 5 inches deep.

These baskets are a product of England. The number of water lily dealers in the USA that handle this product is unknown to me. In 1985, I made one long-range projected purchase of these baskets from Lilypons Water Gardens. Since that purchase, Lilypons decided not to carry this item. Baskets are currently being offered by: William Tricker, Inc. (P.O. Box 31267; 7125 Tanglewood Drive; Independence, Ohio 44131; Ph. (216) 524-3491 or 3492)

Besides the sizes mentioned earlier, a large round basket, 9 inches by 5 inches deep is also available. The Tricker organization is very excited about adding baskets to their product line and is looking forward to helping ICPS members with their needs (Burk 1989). For overseas members, write to: Blagdon Water Garden Center Ltd., Church Street, Blagdon, Bristol B518-6RZ U.K.

Now one can purchase plastic baskets from department stores at a lower cost, but durability due to composition (less rubber) might be questionable, resulting in more expense in the long run. Another CP grower is currently experimenting with another type basket in a modified system. In perspective, this tool is available in one way or another.

Cold Storage

Pantries and basements are poor areas to keep your plants during dormancy due to the uncontrollable fluctuation of temperatures. This factor will cause disturbed and inadequate rest as well as premature growth starts which the northern grower must avoid at all costs until the right moment to safely move outside.

I take my baskets of plants to a local business that provides ice and cold storage for the community which is agricultural-industrial. You rent space as needed, floor or rack. Once inside, you can see anything from crates of apples or string beans to kegs of beer. I also spotted pots of small shrubs on racks one year. The storage building is fairly large and separate from the ice house and is kept cold at a constant 33°F. all year. This building is lit all day, but to a degree that plants do not benefit. An excellent air circulation system is in constant use. With back-up equipment in case of breakdown, I can sleep well at night.

I have never had any of my plants disturbed. This type of storage gives my plants a steady, semi-hard and safe dormancy for a period of five months at a cost of $20.00 per month per 8 foot section.

The availability of cold storage should be determined before committing oneself to this type of cultivation, for it is a critical feature. Shop in the yellow pages and meet personally with the manager. Ask questions about their operation and talk about your needs. They are very understanding and cooperative.

During the formative stages of this system of cultivation, I heard the tragic story of a member a few years earlier losing a very large collection of *Sarracenia* to voles (field mice) eating the whole rhizomes of plants while wintering over under mulch. I felt this was not a safe practice to start and growers who are planning on using mulch or those who have been doing so are taking a BIG risk. I just received another sad report of this happening during the winter of '90-91.

"Seasonal Bog"

This structure is basically a customized peat bed, designed and reserved solely for the cultivation of southern *Sarracenia* in baskets.

The bog can be of any length and configuration desired so as to blend in with your landscape, but a width of 36 inches should be maintained. This size is wide enough to accommodate three large lily-type baskets (though intermixing of sizes will probably occur), and narrow enough to install baskets with ease at arms' length from the "front side" of the bog. The periodic maintenance of plants and bog (Hummer 1979:78; Schnell

1976:103, 111; Slack 1980:198), plus the desire to collect plant data also makes this width workable.

The sides of the bog should be vertical so baskets (which are tapered) can be placed right against the edge and have space for buffering agent (peat) to completely surround them.

A bog depth of 12 inches was used. The minimum depth of 8 inches suggested by Hummer (1979:78) is not suitable for basket use. Baskets should sit on an adequate amount of buffering agent (peat) and, if aeration of baskets is needed (to avoid rhizome rot) due to heavy rain flooding, the water table can then be lowered to the base of the largest baskets and still have a water reserve. I envy those growers who have acid water on tap, and an 18 inch maximum bog depth suggested by Slack (Slack 1980:190) to reduce frequency of watering would seem favorable and easy to manipulate, but the overwhelming majority of CP growers are faced with collecting rain water in some form of cistern. Acid water is at a premium. I felt that for the system I was trying to achieve, the extra 6 inches of peat and water was excessive and could compound some problem situations.

To saturate a large volume of peat when setting up your bog can heavily tax one's water supply. More water is then needed to establish a water table, raise it to the desired level and then hold it there daily. Faced with this operation at a critical growing time can make one's hair gray if you run out of water. I assembled my bog in the fall season and added wet peat. This allowed the peat to age over winter and I did not need any more water until spring.

When I first envisioned this system of cultivation, I wanted to avoid the unnatural and unsightly use of lath or cloth shading. Therefore, the system had to be successful in an average back yard location subjected to long hours of full sun and heat with no mid-day shade. I agree with Hummer (1979:78), that *Sarracenia* need plenty of sun for robust growth. The more the merrier.

After evaluating my property for the best morning sun location (Schnell 1976:109) and taking notes on shade patterns for orientation, the best location available was too limited in space and hours of sun due to a Mimosa tree. By a streak of luck, the Mimosa tree died over winter, which upset my wife but suddenly gave me enough space and light to work with. The area receives full unobstructed sunlight from sunrise to about 4:30 P.M. daily, year around.

These environmental factors influenced my concept on bog plumbing. Due to a high evaporation rate (Slack 1980:190), and the need to conserve as much water as possible, drainage holes (Schnell 1976:110; Slack 1980:190) were not incorporated into the bog system.

After two years of monitoring bog water tables, the rate of evaporation averaged 1 inch loss per day ±1/4 inch.

Heavy rain flooding occurs frequently in the wild and will not harm *Sarracenia* if duration is brief. Periodic flooding is also beneficial to your bog in that it helps flush baskets of accumulated salts and balances the pH of compost. I also welcome flooding because of the large amount of water collected.

Actually, it will take a lot of steady rain, possibly over a day or more depending on how heavy, to raise the water table to a flood level reading of 12 inches. Unless storms persist, the water table will drop at a safe rate. If flooding persists more than a day due to continued storms, then draining is advisable to be safe. I have done this only four times in five years.

NOTE: a 1 inch rise of water table does not correspond with 1 inch of rainfall.

To accurately monitor the amount of water in my bog, I crossed over to the tropical fish trade and purchased a section of 3/4 inch diameter rigid plastic tubing, a section of 3/16 inch diameter rigid plastic tubing and a plastic strainer used to keep fish from being sucked into an aquarium filtering system.

The plastic strainer was pushed over one end of the 3/4 inch diameter tubing (if not a tight fit, glue it). I then measured from the end of strainer an overall length of 14 inches, made a mark and cut off (Figure 2B). The length was determined by 12 inches of peat, 1 inch of live sphagnum ground cover with 1 inch left over to extend above moss. The 3/16 a hand trowel. Recheck the depth again from the top of the nail head; this is now your starting point. As you excavate, insert nails 1 foot apart along the bog wall with the nailheads leveled using a spirit level. By the time you end up back at the starting point, you will have a level bog. The finished excavation should be neat, with all stone holes filled in, roots trimmed off and a flat floor (Figure 4). If you want a walkway around your bog, it should be measured out, the sod cut and the soil removed to the desired depth at this time. Back during the planning stage, I determined the bog circumference by laying a piece of string on the graph paper following the contour, then measuring the string to scale. I then ordered from General Foam Plastics Corp. Norfolk, Va. ph. no. 1-804-857-0153, (the makers of 18 inch high children's swimming pools), the amount of plastic wall needed. These walls are sold in 25 foot lengths. After receiving the wall material, I cut the width down to 13 1/2 inches on a table saw and then peeled off the clear sheet of designs.

To fasten wall sections together, I overlapped the ends 3 inches and, with a thin hot nail, I perforated three double sets of holes 1 1/2 inches in from the edge while laying sections for perfect match. The sections were carefully wired together with the twisted wire ends on the outer side of the panel. This long finished panel is not cut to its correct length or the ends united until after installation. This plastic wall subliner is used to protect the main bog liner from puncture holes and, at the same time, providing a lip to prevent lawn soil from washing over and into your bog soil (Slack 1980:192).

To assemble the bog, the long subliner was stood up against the bog wall with the unconnected ends at one end of the bog. The subliner was temporarily held in place with bricks. Wide duct-tape is placed over the inside seams of the subliner where connected to protect the main liner. On the dirt floor, pieces of window screen, tar paper shingles, or other material, is carefully laid down to protect the main liner from holes caused by boring *Cicadia sp.* (17 yr. locust) or similar pests. On top of this covering, a 1/2 inch layer of wet newspaper is neatly placed, fitting all corners and forcing the subliner against the bog wall as you go. After a few years, the paper will decompose into a watertight substance called gley, so that even if the liner is punctured, your bog will retain water. The main liner, which is 6 mil. sheet plastic obtainable in 100 foot rolls from a local agricultural supply store, was cut to a width four times that of the bog and 10 feet longer, due to the bogs' curved shape. This liner was carefully laid inside the bog and at the end of the bog with the connected subliner, the liner was laid over the subliner lip and temporarily held in place with snap-on clothes pins. The liner was fitted to the bog for a short distance and held in place with more clothes pins. I did

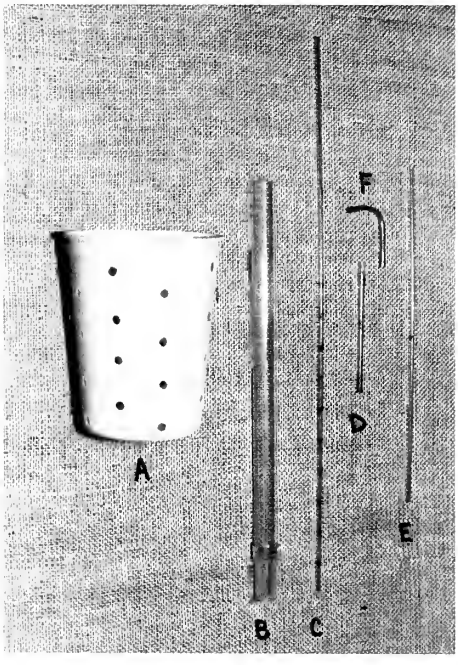


Figure 2. Plumbing Kit

this work while kneeling in the bog. My son then dumped 6 inches of wet peat into that area while I packed it down good. The process of folding pleats, fitting, fastening with clothes pins and dumping peat, continued until the opposite end of the bog was reached. By this time, the subliner has conformed to its finished dimension. Allowing for a 3 inch overlap, excess subliner was cut off with a sharp knife and the ends connected together as mentioned earlier. The main liner was then fitted into this area and peat added. The whole bog was then filled with wet peat to within 1 inch of the lip. Allowing 6 inches of overhang, all excess main liner was trimmed off and the clothes pins removed (Figure 5). At one end of the bog, about 6 inches from the edge, I dug a hole with my hand to where I could see the main liner. I stood the water meter (Figure 2B) up, making sure it wasn't resting on a fold, I then replaced the peat. If you want the white subliner lip to blend in with the soil, you can buy clear primer spray from an auto bodyshop, then use Krylon® brown spray enamel to cover up. Also at this time, your walkway should be filled in to within 2 inches of the bog lip with the material of your choice. The bog is now basically installed (Figure 5).

The remainder of the main liner was trimmed off the following spring after everything settled, at which time the excess main liner is folded over the subliner lip and held in place with snap-on plastic molding strips. The excess main liner was then closely trimmed off. This snap-on trim is not only necessary to hold the main liner in place, but also prevents insects from crawling between the two liners. An unidentified insect made a flat white cocoon on the main liner, underwent metamorphosis and chewed through the liner and into the peat, leaving holes as large as 1 inch in diameter in the liner. I suddenly started using large amounts of water. I exposed a section of liner, suspecting a puncture of some sort. In the spring of 1989, I replaced the main liner. Of all the calculations and reasoning needed to make this method of cultivation work, I had to stumble over something like this!! I was upset with myself with every shovel full of peat I had to remove. So, get that trim on early in the spring. The snap-on molding was also purchased from General Foam Plastics Corp.in 25 foot lengths, and is normally used for pool liners. Order green color molding.

The total cost of constructing the "seasonal bog" portion of my wetland complex, including plumbing material and 7 bales of peat, but excluding baskets and sphagnum moss ground cover (which is an annual expense), was $180.00.

I consider the START of the growing season as that time when it has been determined that all danger of frost has passed, thus allowing plants to be placed in the "seasonal bog". In my area, Mothers' Day is when the local people safely set house plants outside. I've been regulating my spring activities around this local tradition.

The transition of plants from conventional containers to baskets and/or replanting is started in the spring (Slack 1980:198, Schnell 1976:113) four weeks before the growing season begins. Only those plants that need to be converted over or replanted are removed from cold storage at this time; the remainder stay in dormancy. The plants are brought home, watered as in dormancy, (using water that is of room temperature), then placed in indirect light. In response to five months of constant semi-hard dark rest, within 48 hours after bringing plants home, all growth points usually show new growth. I wasn't prepared to experience this aggressive reaction the first time. Conditions are not changed the second week. At the beginning of the third week, replanting is performed. With a modified basket, as described earlier, "bridals' veil" is laid inside and moist compost added. I prefer a compost consisting of a peat perlite ratio of 2-1. Taking the plant rhizome and more compost, I form a mound about an inch above the basket rim (to allow for settling) and the top surface of the rhizome completely exposed. I then carefully firm compost against the rhizome, covering any exposed roots. Excess liner is trimmed off and a plant identification tag is inserted into the basket. A log is kept, listing all plants and the dates that they were put up in baskets. The plants are then set on the lawn and watered lightly with the rose of a can

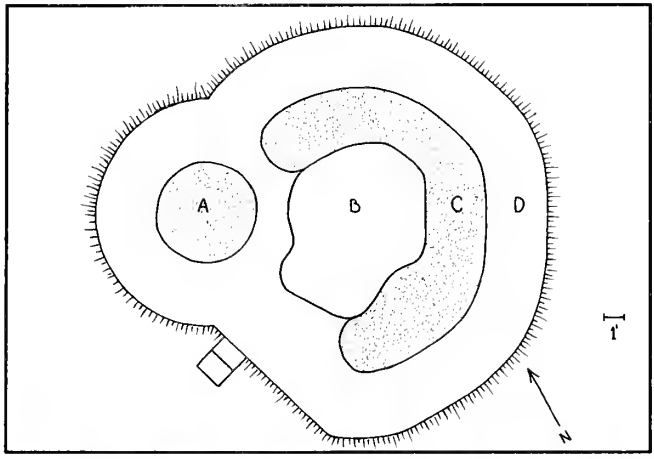


Figure 3. Diagam of wetland complex


Figure 4. View of excavation for wetland complex in September 1985

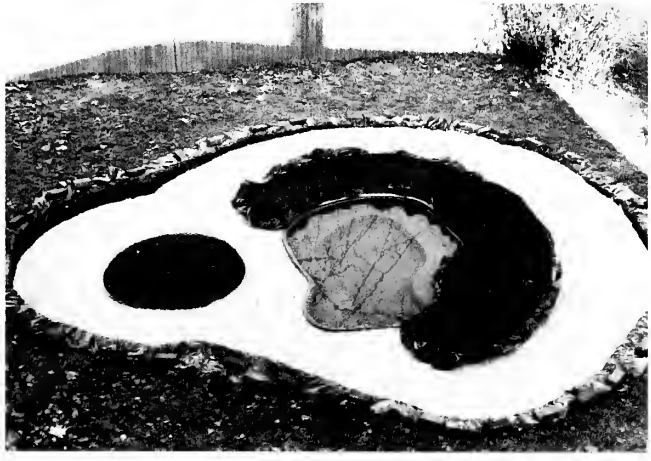


Figure 5. View of wetland complex after installation in October 1985

and exposed rhizomes are misted with Benomyl®, then placed on our porch for the next two weeks, making sure that they receive only indirect light and the compost does not dry out. This period of porch time allows plants to adjust to fluctuations of day and night temperatures, air currents and photoperiod. The transition from dormancy to a growing cycle is a VERY important element (Schnell 1976:97) and should not be bypassed. If during this time there's a last minute drop in temperature, foliage will not be harmed by frost because of protection by the porch roof.

It must be said, that because of the time involved adjusting plants from pot culture to baskets and then to the "seasonal bog", size and shape of the first pitchers will be sacrificed the first year.

Every spring about two weeks before the start of the growing season, I remove by hand any accumulated debris and all remnants of sphagnum moss from last year's ground cover from bog surface. With an "onion rake", I carefully till the upper 4-6 inches of peat, watching not to snag the sides of the main liner. The peat removed the year before during installation of baskets is once again added to fill in all depressions. The peat is then carefully raked level with a regular iron rake and, if needed due to settling, new wet peat is added to within one inch of the bog lip. At this time, to insure that the water meter (Figure 2B) is resting on the bottom of the bog, I twist and push down on it. The gauge tube (Figure 2C) is then dropped in. At the opposite end of the bog, I insert the watering pot (Figure 2A) to its rim. A piece of flat bark is then laid on top to conceal the pot and act as a cover. You may or may not have a water reading on your gauge tube, but a high water table is not needed at this time.

On the Saturday closest to the first of May, the plants which are in baskets are brought home from cold storage. Upon arrival at home, baskets are set on the lawn and watered well using the rose of a can. Exposed rhizomes are misted with fungicide and baskets are left to drain a few minutes. Plants are then placed on our porch for the remainder of the week, making sure that they receive only indirect light and that compost does not dry out. The plants are kept on the porch for at least seven days if the weather has been abnormally warm for a while or they remain on the porch until the Saturday closest to Mothers' Day if you fear the chance of a last minute frost. Both types of transition periods have produced 3 inch pitchers and/or 5 to 6 inch flower scapes during this time.

On the day that my plants are to be placed in the "seasonal bog", I mentally arrange the plants so that the smaller varieties are toward the "front" of the bog for best observation and east of the taller pitcher plants (Schnell 1976:110, 111) so that they are not shaded and deprived of valuable morning sun.

Starting at one end of the bog, I take an empty basket of the size that is to be placed into the bog and lay it on the surface upside down in the location desired and trace the outline with the tip of a small hand shovel. I proceed to dig a hole to the shape of the basket, putting the peat that is removed into a bucket which is later stored in a garbage bag. The empty basket is placed in the hole to judge proper fit. Once the basket sets in the hole with the rim level with the surface, I look into it to see if the peat is against all sides. The bottom of the basket should be sitting firmly on peat also. If I see any open spaces or pockets where peat is not up against the basket, I add peat to that area and then replace the basket and recheck. Any air pockets left unfilled will have a negative effect on the proper hydration that the plant needs. Once I'm satisfied with the inside of the hole, I remove the empty basket and insert a planted basket. This process is repeated across the bog for every plant (Figure 6).

As I work my way across the bog, I carefully lay an inch of live sphagnum moss around the plants (Figure 6), making sure NOT to cover any growth points or rhizomes, for rot may occur. The moss not only conceals the baskets but also serves as a protection for the baskets from damaging ultra-violet light rays, at the same time preventing peat from splashing onto foliage during a rain. Hummer's (1979:78) suggestion that one

Figure 6. Detail of 'planting'

need not cover the bog surface completely because the moss will grow cannot be applied to a temporary "seasonal" bog, where results are short lived but needed immediately. In areas of my bog that cannot be planted for want of plants, I don't bother covering with moss. Isolated plantings are still surrounded with moss which gives the bog a special effect.

On initial exposure to full sun after plants are installed in the bog, there will be a moderate sunburn experienced, which never gave me reason to be concerned for my plants. York, Pennsylvania receives only a 50% average per year of sun bright enough to cast a shadow, as compared to a 65% average for the Green Swamp of North Carolina (Antolini 1978:36). As a result, I haven't attempted to protect nor do I recommend the shading of any southern *Sarracenia* for coloration of foliage will be sacrificed particularly the all red variants. *S. purpurea ssp. venosa* shows no negative effects from this unobstructed exposure level as suggested by Schnell (1976:106,111) to avoid in a more intense southern location.

Upon completion of plant installation and the laying of moss, which is done on a Saturday because of the workload involved, I proceed to fill the bog with water and establish a water table. I'm currently operating my system with approximately a 7 inch water table that is read off the gauge tube. This puts the water table approximately 5 inches below the plant rhizomes, which seems to provide a good degree of hydration. I plan to experiment cautiously with a higher water table in the future. For the size of my bog, I usually start with 32 gallons of water, which is added via the watering pot. When adding water, it takes about an hour or so for it to seep through the peat and reach the water gauge at the far end before an accurate reading can be made. Once the water has stopped moving up the gauge tube, more water is then added if needed to reach the level desired. Once this is accomplished, your system is in operation.

Besides the raising of the water table due to rain and the average daily loss of an inch of water caused by evaporation, I check daily and, if needed, I add water to maintain at least a 7 inch water table until all pitchers have completely opened; for I agree with Schnell (1976:98,106) that an even, constant, high water table is critical to achieve maximum growth.

To avoid the time consuming task of adding water everyday, waiting to see how far it moves up the gauge tube, then adding more water if needed, on a day that watering was needed, I took a reading, poured two buckets (plastic) of water into the watering pot, waited two hours to make sure that the seepage was complete, took a second reading and calculated out how many buckets of water would be needed to raise the water table one inch. As a result of this test, I can come home from work, take a reading and, if the water table is down one inch, I just add three buckets of water and go about my business without rechecking.

For the first two weeks, to accelerate the establishment of the sphagnum ground cover, every other day that I must add water, I pour that day's required amount over the moss using the rose of a can. Then, for the remainder of the season, once a week (if it doesn't rain), I'll take that day's water and pour it on all the plant rhizomes (using the rose of a can) as a treat. I do not consider this a necessity though.

Besides the required watering, once a week I mist my plants (Schnell 1976:106) using a heavy duty pressure sprayer as advertised in garden and seed catalogues. This is performed solely for the purpose of keeping the pitchers' esophagus moist, which is needed for the plant to absorb nutrients from the biomass. I aim the mist directly, but moderately, at the pitcher openings. This application is helpful when the air might become periodically dry. I also direct a shot of water at newly opened *S. purpurea* pitchers, to give them a head start for they require a high humidity (Schnell 1976:106).

All this watering might seem burdensome, but actually I spend about a half hour a day at watering the whole backyard complex. This is always done in the cool of the evening, never in the hot direct sun so as to avoid plant shock and leaf scorching.

Once all pitchers have opened, I am not fussy about maintaining a daily 7 inch water table. During the drought of 1987, I deliberately cut back on water to conserve, which meant that if I had a water reading of 6 inches I was good for six days. I once stopped watering with a 7 inch reading and didn't add water for fourteen days (during which time there was no rain) with no ill effect to the plants for the peat was still damp and cool, which is important for the plants' roots. This proves excellent for making it possible to go on vacation by raising the water table to a 12 inch reading before going on vacation for two weeks. After returning from vacation, water is added until a 7 inch water table is achieved, which is why I felt that an 18 inch deep peat bed, as mentioned earlier, was excessive. If one allows the bog to go dry, it would require 13 inches of water instead of 7 inches to recover.

At least once a year (usually in the fall), the pH and TDS (total dissolved solids) of your bog should be monitored. With most *Sarracenia* species growing in an acidity range of 4.5-4.7 (Wherry 1929), it is my opinion that a higher pH level would have a negative impact on the plants' metabolism and disease resistance system. As a result, I consider the bog peat useless once it reaches 5.0 (5 times weaker than 4.5) at which time it should be replaced. I use the La Motte model P-BEG (code 2105) pH test kit (Stahle 1987:8). As for TDS, Stoutamire (1972:6) reports that "mixtures in which *Sarracenia, Drosera* and *Sphagnum* mosses grow", range from 20-40 uMho; 40 micromhos equaling 28 ppm. To avoid a high level of toxicity, I'm following Schnell's (1976:97) recommendation that TDS "should be less than 50 parts per million (equivalent to 100 micromhos—)". A local nursery man has been kindly testing the TDS for me but one can purchase a small test kit from La Motte. I siphon my two samples of water from the bottom level of the bog using a length of rigid tubing without an elbow (Figure 2E). Yearly test results for both pH and TDS are entered into a log so I can monitor the deterioration in bog quality. Since tipping (Schnell 1976:101) is impossible as a means of flushing out toxic levels of salts, one could siphon all the water out of the bog after a heavy rain to possibly add another year's life to the peat. I will most likely change the peat. It is my goal at this time to maintain at least a seven year repotting cycle. This time span will be determined by the condition of the bog peat and when it will need to be replaced based on the levels of pH and TDS, whichever becomes unacceptable first. I'm counting on the bog peat to last at least five years, if not longer. The bog peat should always be replaced first which, in turn, will buffer the planting compost by seepage, thus adding another year to the usefulness of the compost. If this sequence would be reversed, the new planting compost would be wrongfully affected.

One manifestation of "season bog" cultivation resulting from total darkness and unfluctuating temperatures for five months of cold storage is that all my southern *Sarracenia* have the same flowering date peaks. Admittedly, this observation was

made based on a meager sample of two purchased plants representing the Gulf Coast (30° N. Lat.), but the remainder consisted of purchased or collected representatives of all species commonly colonizing the North Carolina coastal plain (35° N. Lat.). Regardless of latitude or species, a two year average of flowering date peaks was fifteen days after baskets were installed in the bog as opposed to Schnell's comparison of greenhouse and native habitat flowering date peaks (1977:168).

First year cultivation at 39°58' N. Lat. was also confusing to some plants because of day length and temperatures, resulting in three *S. flava* clones producing flowers on four to six inch scapes the first week of September. This manifestation didn't occur the second year once the plants became regulated.

Of two *S. flava* hybrid clones salvaged (Schnell 1976:113) within two feet of each other from Brunswick Co., N.C., one of these clones has produced a second crop of pitchers of normal height in early September since being in cultivation.

Come mid-September, I discontinue adding water to the bog and let the weather control the water table in order to prepare the plants for the dormancy period ahead (Schnell 1976:106). At times, there may not have been a readable water table but my bog has never dried out completely.

When I get up in the morning and have to scrape ice off my windshield (which has been occurring in my area about mid November), it's that time for me to prepare my plants for cold storage.

In the evenings, a couple of baskets at a time are "lifted" from the bog and set on the lawn to drain. Afterwards, they are brought into the house to be cleaned up. All peat is brushed off the outside of the baskets and any roots growing through the basket are carefully trimmed off. Directing my attention next to the top of the basket, all moss and grasses are carefully removed.

When it comes to prepping *Sarracenia* , I try to retain as much foliage as possible (Slack 1980:198), at the same time thinning out foliage and eliminating the possibilities of insects being carried over to the next season, by removing all completely formed tubes. As a result, on those species that produce winter phyllodia, the phyllodia are retained (Schnell 1976:111) and all tubes (even if still green) are carefully trimmed off as close to the rhizome as possible with a pair of manicuring scissors. Any dead ends of the remaining foliage are trimmed off. For those species that do not produce phyllodia and whose pitchers can survive in protected areas (Schnell 1976:34, 37-44), I trim off half the foliage from each growth point, preferably saving incompletely formed tubes.

S. purpurea ssp. is truly an evergreen species. Tests conducted by me during a four year period confirms that this species stores its nutrients in its foliage and to cut off the foliage will set the plant back causing a very slow recovery, if not death. The following season, only the completely dead foliage should be removed from this plant. I instinctively treat *S.* x *catesbaei* with the same respect. For those growers that are custodians to other *S. purpurea* hybrids, it might be of value to consider this aspect.

Once all unwanted foliage is removed, compost is repacked up against the rhizome to eliminate any air spaces underneath. The rhizome is then misted with fungicide. The plants are now ready for storage. They can be placed in cold storage as they become ready or kept in a cool basement and taken into storage at one time, which I normally do the first of December. This two week period has no negative effect on my plants.

So that I don't forget, the gauge tube (Figure 2C) is brought indoors at this time so as not to freeze and break.

The moss removed from the tops of baskets, and that which remains on the bog surface, is not kept for the following year even though replacement involves some cost, because of the grasses, etc., that sprout up during the summer which can give your bog a natural effect but are very difficult to separate from the moss for the next year. If this moss is reused, these weeds, would get out of control. I consider the time I spend

around my wetland complex valuable and, if I spent unnecessary time weeding, that would rob me of time spent working with and enjoying my *Sarracenia*. Therefore, I only take time to weed out my northern bog of unwanted growth. The ordering of sphagnum moss is, other than cold storage fees, my only annual overhead. Moss is ordered early in the Spring from: Mosser Lee; County "O" & I-94; Box 437; Millston, Wisconsin 54643-0437.

When renting a section of cold storage rack, make sure that the space you obtain is not located right below the cooling system. In order to maintain a 33° F. temperature at the far end of the building, a colder air output is needed and,if plants are right below the cooling unit, they will freeze. This happened to me during the third year of dormancy when I failed to check where my reserved space was located. Then, three weeks after storing plants, all the bulky stock above my plants was removed from storage, forcing all that cold air down through the racks and onto my plants. So reserve space ahead of time to insure available space and a safe location.

ALL *Sarracenia* can take a mild freeze, but the period of transition from storage to bog is a sensitive one. You do not want to take your plants out of storage and shock them by putting them immediately into the hot sun. At the same time, you do not want to keep the plants in a protected area longer than necessary where you can retard and deform the foliage because of inadequate hydration and light. As mentioned earlier, plants will come out of dormancy quickly, but this freeze of 30° F., which I consider mild, caused an additional two weeks for the plants to recover (causing me frustration) because of heat. I lost six plants to stress from forcing them too hard; so beware.

On the day my plants are to be transplanted to cold storage, they are watered and sprayed with fungicide as in dormancy. Contrary to popular beliefs, if you keep *Sarracenia* outside untiI the first hard frost, which gives your plants' natural "antifreeze" system a chance to activate (Mellichamp 1981:52), your plants can be stored in wet compost without fear of rot. When I hear of growers losing plants because of rhizome rot during dormancy, the problem can usually be traced to not allowing plants to remain outside long enough for their antifreeze system to kick in and/or plants are stored in a basement which is not cold enough (33-35°F) and/or the temperature is not kept constant, which will negate the plants' "antifreeze" (dormancy) system. My baskets are not allowed to dry out and, in fact, are kept wet at all times. Potted plants are also kept outside until frost and then stored wet with no negative effects.

My maintenance program for dormancy is as follows: Every Saturday morning, I check my plants to make sure that the baskets didn't dry out. Because of the air

circulation system, you might have to go twice a week until you get familiar with the evaporation rate. I take with me a couple of plastic containers of water, a spray bottle of fungicide and a small plastic watering container with a curved spout without a rose. I first spray with fungicide if I see any white fuzzy growth starting on top of the compost or rhizome. Even though this might occur (which it did one year) with a weekly spraying to eradicate fungi as needed, my plants have never been affected. Water is carefully applied to the compost, avoiding contact with the rhizome (which can cause fungus to start), by rotating the basket as I water. Only fungicide is allowed to come in contact with plant rhizomes.

Going into my sixth growing season, an evaluation of this system in relationship to the plants can be made.

Due to a high acid level (pH 4.0) as opposed to pot culture leaching, coloration is improved (provided ample sun is received). Plant growth is very aggressive causing multi-growth point clumping reminiscent of Green Swamp plants, as one grower observed. Even Gulf Coast plants that normally produce but one growth point branch off creating large clumps. Since this system of cultivation was started, I have had the opportunity to see a large number of "southern" beds in operation, basically following Carroll's (1982:84) format. With one notable exception, coastal plain mix was substituted for peat in all cases. I have not seen the robust growth that I am accustomed to in my system. This is most likely due to differences in hydration. The root systems are stronger and triple in mass than those produced in sand beds.

On the other hand, *S. minor* does only fair in my system. This requires more work. I also have not achieved the pitcher height yet that I would like to see in some species. This might be due to not subjecting my plants to a water table higher than 7 inches at the very start of the growing season which could possibly force the plants too hard (Schnell 1976:97). More work needs to be done concerning this. Putting aside the six plants that were lost because of MY failure to insure that the plants would not freeze as mentioned earlier, keeping within the framework of the system outlined, I have not been able to keep *S. purpurea ssp. venosa* alive through the third dormancy cycle. This is a double-edged sword for me because the foliage MUST be retained from one year to the next, causing the foliage to cover the rhizome resulting in poor ventilation. The plant cannot be properly sprayed with fungicide and then dies. This is only a preliminary evaluation. I have not grown *S. psittacina* yet but I can see this problem possibly occurring with this species also.

From the beginning, it has been my intent to create a good system of cultivating *Sarracenia* for the northern grower. Some of the many tests that I have subjected my plants to could be called "reckless". In come cases, I have done the opposite of what I had read not to do. These plants are not as fragile as I earlier understood. There always will be tests to perform, manifestations to evaluate and possibly changes for improved cultivation, but then again *Sarracenia* has the ability to consume one's curiosity. Hopefully, this system will open the door for more northern growers.

Literature Cited

Antolini, Holly Lyman (Ed.). 1978. Sunset Homeowner's Guide to Solar Heating Lane Pub. Co., Menlo Park, Calif.

Bell, C. Ritchie. 1976. "Foreword" in D.E. Schnell, Carnivorous Plants of the United States and Canada

Burk, Amy. 1989. Personal communication. William Tricker, Inc., Independence, Ohio

Carroll, Bill. 1982. News and Views. CPN 11(4):84

Heritage, Bill. 1981 Ponds and Water Gardens. Blandford Press Ltd. Poole, Dorset BH15 ILL U.K.

Hummer, John H. 1979. Are You Ready For A Bog Garden. CPN 8(3):78,95
Koopowitz, H. and Kaye, H. 1983. Plant Extinction: A Global Crisis. Stone Wall Press, Inc., Washington, D.C.
Mellichamp, Larry. 1981. Horticulturists' Corner. CPN 10(2):52-53
Schnell, Donald E. 1976. Carnivorous Plants of the United States and Canada. John F. Blair Pub., Winston-Salem, N.C.
_____. 1977. Infraspecific Variation in *Sarracenia rubra* Walt: Some Observations. Castanea 42:149-170
_____. 1978. *Sarracenia flava* L. Infraspecific Variation in Eastern North Carolina. Castanea 43:1-20
_____. 1980. A Photographic Primer of Variants of *Sarracenia flava* L. CPN 9(2):41-44
Slack, Adrian. 1980. Carnivorous Plants. MIT Press, Cambridge, Mass.
Stahle, Jerry Sr. 1985. The Cataloging of CP Stations. CPN 14(2):51-54
_____. 1987. News and Views. CPN 16(1):8-9
Stoutamire, Warren. 1978. Monitoring Salt Levels in Bog Potting Soils. CPN 1(1):6
Swenson, Allan A. 1977. Cultivating Carnivorous Plants. Doubleday Co. Inc., Garden City, NY
Wherry, Edgar T. 1929. Acidity Relations of the *Sarracenia*. Journal Washington Academy of Science 19:379-390

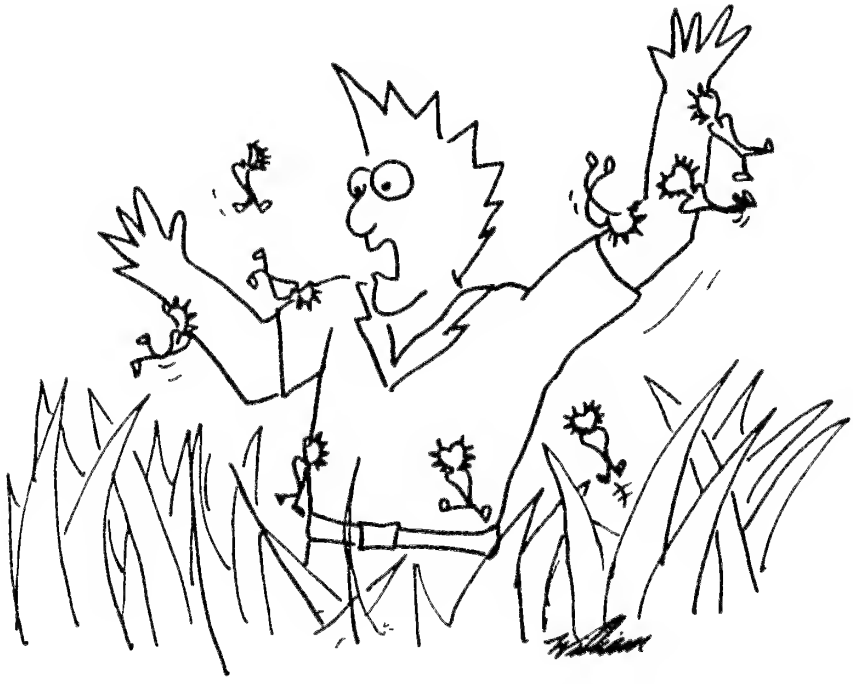
UPCOMING ARTICLES

- A Slow Sunday at the Greenhouse
- Mechanisms of Trap Movement II: *Aldrovanda*
- CP in Ireland III: David Moore
- A Letter from Sierra Leone, re: *Triphyophyllum peltatum*
- CP Tour of South America
- *Nepenthes* at Longwood Gardens - an Update After 12 Years

TIDBITS

BY

BILL STRAND



UNWITTINGLY, PROF. LUIS DISCOVERS

Dionaea muscipula ssp. *piranhadae*